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1 Introduction to anaesthesia in exotic species

INTRODUCTION

Exotic animals are popular pets, and often present to the veterinary practice for evaluation and treatment. These species are varied anatomically and physiologically from the more commonly presented species. These differences will affect how the patient responds to handling, illness and anaesthesia.

WHY IS ANAESTHESIA NEEDED IN EXOTIC PETS?

Anaesthesia of animals may be necessary for two main reasons: to cause immobilisation to allow examination or performance of minor procedures (for example phlebotomy), or to perform surgical procedures humanely by causing loss of consciousness whilst providing analgesia, muscle relaxation and amnesia. The presence of each of these factors is dependent on the anaesthetic used, with local anaesthesia not causing unconsciousness and some general anaesthetic agents producing relatively little muscle relaxation. The requirements for these facets vary between cases and the clinician must consider what is necessary for the animal in question before selecting an anaesthetic regime.

Anaesthesia is required for many varied procedures in exotic pets. Certain species cannot be manually restrained without injury to handlers or stress to themselves, and sedation or anaesthesia is required even to perform a clinical examination. In other more amenable species, anaesthesia may be required for investigative procedures or surgery. If surgery is to be performed, analgesia should be provided. Analgesics will be briefly discussed, principally in the context of an aid to anaesthesia.

PRE-ANAESTHETIC ASSESSMENT AND SUPPORTIVE CARE

Inadequate or inappropriate husbandry often predisposes exotic pets to disease and an important part of the pre-anaesthetic evaluation will involve taking a thorough history of the animal's current and previous diet and environmental conditions. A complete history and understanding of species' requirements are vital in these pets as clinical examination before anaesthesia may be difficult (for example in very small rodents) or limited (for example due to the chelonian shell). Later chapters will discuss husbandry conditions in various species that may predispose to or cause diseases, for example those affecting the immune and respiratory systems.

A clinical examination should be performed, if possible, with minimal stress to the patient. At this stage, a weight should be obtained, to enable accurate dosing of drugs and subsequent monitoring of body condition. Many species become stressed when restrained, and high circulating catecholamines may predispose to cardiac arrhythmias. Pre-anaesthetic history taking and clinical examination will allow the clinician to form a picture of the patient's health status, in order to identify any increased risks pertinent to the individual pet. Even if none are found, the benefits and risks of anaesthesia should be explained to the animal's owner. Written consent should be obtained for the procedure, as most drugs are not licensed for use in exotic animals (this will vary between countries). The veterinary surgeon should also advise the owner that the duration of many drugs (including analgesics) has not been verified experimentally in many species, but is based on clinically perceived durations of action.

If possible, a small blood sample should be obtained before anaesthesia to assess the patient's packed cell volume (PCV), total protein, blood urea nitrogen (uric acid in reptiles) and blood glucose (Heard, 1993). These parameters will allow assessment of the animal's hydration and nutritional status. Dehydration and malnutrition are common in exotic pets presented to the veterinary surgeon, and it is often prudent to postpone anaesthesia while fluid and nutritional support are provided to stabilise the patient's condition. Although this text is primarily concerned with anaesthesia in exotic pet species, much of the success of

anaesthesia in these animals relates to provision of sufficient care in the perioperative period. Information is, therefore, provided on nursing and supportive care, including basic hospitalisation techniques, fluid and nutritional support.

ANAESTHETIC EQUIPMENT

Equipment for use in anaesthesia varies greatly, the primary requirements being delivery of anaesthetic agent and oxygen to the patient, and scavenging of waste gases. Waste gases contain carbon dioxide produced by the patient, and anaesthetic agents that would cause environmental contamination and potential risks to staff.

Anaesthetic machines

In order to deliver oxygen and anaesthetic gases to a patient, an anaesthetic machine is required. Machines for dog and cat anaesthetics are suitable for use with exotic pets. Oxygen and nitrous oxide can be provided from cylinders stored on the anaesthetic machine, or via pipes from a bank of cylinders in the hospital situation. Flowmeters are usually not capable of accurate delivery of low gas flow rates. Small rodent anaesthetic machines have been suggested (Norris, 1981; Sebesteny, 1971) to overcome this problem, but the flowmeters on most machines can still be used providing a minimum flow rate of 1 L/min is maintained. Calibrated vaporisers are necessary for addition of volatile anaesthetic agents to carrier gases (usually oxygen), and are specific for different agents (Flecknell, 1996).

Anaesthetic circuits

The most commonly used circuit for small animal anaesthesia is the *T-piece* (Fig. 1.1) (Ayre, 1956). This circuit has low resistance and little dead space. The presence of a reservoir for anaesthetic gases, as a tube with or without a bag attached (the Jackson-Rees modification), enables the gas flow rates to be reduced to twice the minute volume. The addition of a reservoir bag enables positive pressure ventilation to be performed. Dead space can be minimised by using low dead space connectors, and minimising space between the animal's muzzle and the mask (Flecknell, 1996).

The *Bain* is a coaxial version of the T-piece circuit, with the inspiratory part running within the reservoir limb (Fig. 1.2). This has the advantage of reduced 'drag', as a single tube runs between the anaesthetic machine and the patient, and the reservoir bag and scavenge are located away from the patient (Flecknell, 1996). For animals weighing less than 10 kg, modifications with a valve and reservoir bag cause too much resistance; however, an open-ended reservoir bag may be attached. This latter modification allows positive pressure ventilation to be performed on the patient. The gas flow rate for a Bain circuit is 200–300 ml/kg/min (Ungerer, 1978), or 2–2.5 times minute volume.

Mechanical ventilators can be connected to either T-piece or Bain circuits.

Magill circuits (Fig. 1.3) can be used in animals weighing more than 10 kg. Circuit resistance is quite high and the dead space within the circuit is typically 8–10 ml (Flecknell, 1996).

The above three anaesthetic circuits are non-rebreathing systems. Closed breathing systems, such as the *circle* (Fig. 1.4) and to-and-fro, utilise a soda lime canister to absorb expired carbon dioxide, enabling rebreathing and recycling of anaesthetic gases. They are often run semi-open, with fresh gas flows of 0.5–1 L/min. These systems are useful for larger animals, as lower gas flow rates are required and

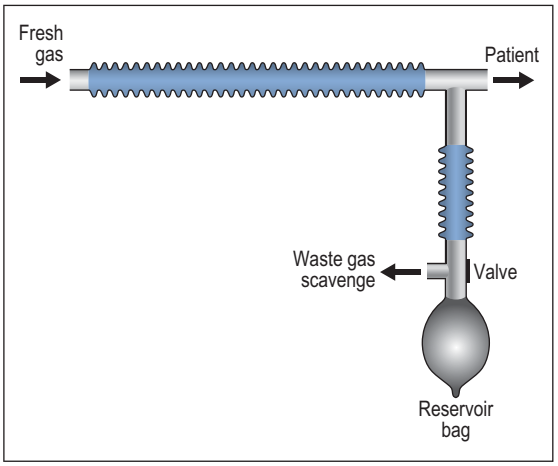


Figure 1.1 • Schematic of T-piece anaesthetic circuit. The addition of a reservoir bag and valve allows intermittent positive pressure ventilation to be performed easily.

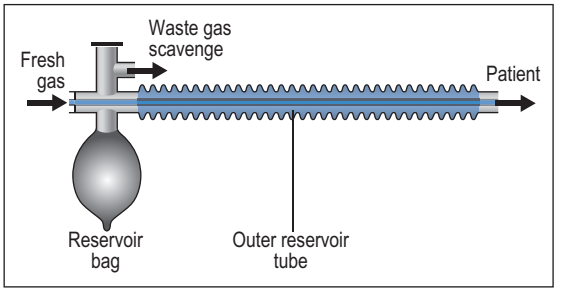


Figure 1.2 • Schematic of modified Bain (coaxial) anaesthetic circuit.

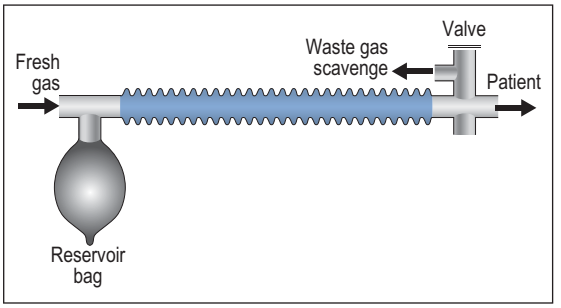


Figure 1.3 • Schematic of Magill anaesthetic circuit.

costs can be reduced as less anaesthetic agent and oxygen are used. However, the valves and soda lime within these systems increase circuit resistance, and they can only be used in smaller animals (less than 5 kg) if mechanical ventilation is used. Nitrous oxide is not used routinely with closed systems, as it may build up and reduce the oxygen concentration significantly (Flecknell, 1996).

Gas flow rates are calculated for each circuit type, and depend on the amount of gas used by the patient. The minute volume is the total volume of gas inspired by the animal in 1 min, and is calculated by multiplying the tidal volume by the respiratory rate. As animals do not inspire continuously, the gas flow rate is usually higher than the minute volume. For example, the flow rate needed may be three times the minute volume for an anaesthetic delivered via a facemask attached to an open circuit when the patient inspires for one-third of the minute (spending the rest of the time exhaling, and pausing between inspiration and exhalation). Non-rebreathing circuits require oxygen flow rates of two to three times the minute ventilation, which is approximately 150 to 200 ml/kg per minute (Muir and Hubbell, 2000).

For many small patients, this flow rate will be miniscule, and the fresh gas flow rate on the anaesthetic machine may not be titratable to this level. For example, a rabbit weighing 2 kg may have a tidal volume of 10 ml and a respiratory rate of 40, and, therefore, a minute volume of 80 ml (10 ml × 40), which requires a gas flow rate of 240 ml/min if using a T-piece circuit. The flowmeter on many anaesthetic machines is not accurate below 1 L/min, so this should, therefore, be used as a minimum setting.

The end of the respiration part of the circuit contains expired gas. Gases within this 'dead space' are re-inhaled by the patient, including high levels of carbon dioxide produced by the patient. If the dead space is large and high concentrations of carbon dioxide are inspired, this will be detrimental to the patient (Flecknell, 1996).

Resistance to the flow of gases, for example caused by valves, within the circuit may also increase the effort required by the animal to move gases during respiration (Flecknell, 1996). This will be particularly significant in small patients that normally have low tidal volumes (i.e. the volume of gas inspired with one breath).

Scavenging is an important part of an anaesthetic system, removing anaesthetic agents safely to reduce exposure to personnel in the practice. This may be performed by connection of waste gases to an active scavenging system, or to activated charcoal for adsorption. Activated charcoal systems are ineffective at removing nitrous oxide (Flecknell, 1996).

Connections to the patient

The use of *induction chambers* to induce small animals has both advantages and disadvantages. Minimal restraint is required before anaesthesia, reducing stress to the animal and potential danger to the clinician. However, most volatile agents are irritant to the airways to some degree, and certain species may breath hold. It is, therefore, advisable to pre-oxygenate the patient before the anaesthetic gas is added to the chamber. It is more difficult to assess depth of sedation or anaesthesia when the patient is within a chamber; this is improved by using clear containers (for example, Perspex®, clear Tupperware® or plastic bottles [Fig. 1.5]).

Ideally, the induction chamber should have an inlet pipe for gases as well as a scavenge outlet. Gases should be scavenged from the top of the chamber to remove that containing a lower concentration of the anaesthetic agent, which sinks below air as it is denser. Where plastic bottles are used to make chambers (Fig. 1.5), the anaesthetic circuit is usually attached to one end; fresh gas administration and scavenge are achieved through high flow rates displacing gases within the chamber. In most systems, there will be environmental contamination when the patient is removed from the chamber, as volatile anaesthetic agents are released. To reduce the risk to staff, there should be good ventilation (but not open windows through which patients could escape!) within the room to allow escape of these agents. Double chamber systems are available and enable removal of anaesthetic gases before the chamber is opened (Flecknell, 1996).

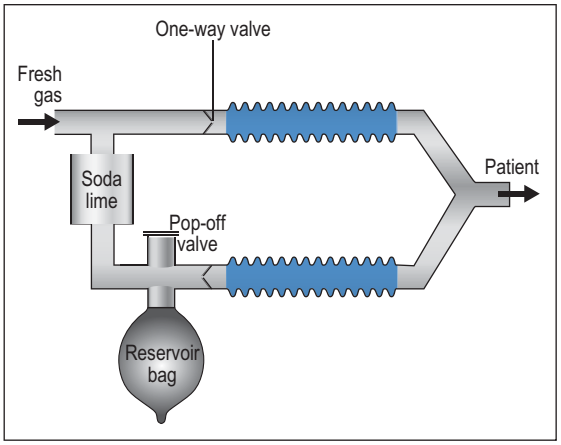


Figure 1.4 • Schematic of circle anaesthetic circuit.

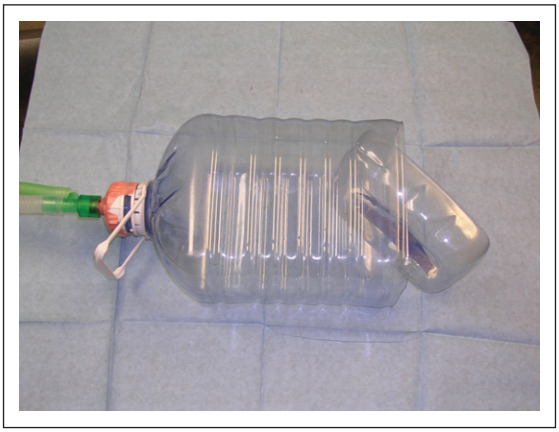


Figure 1.5 • Plastic bottles can be adapted for use as induction chambers with small animals.

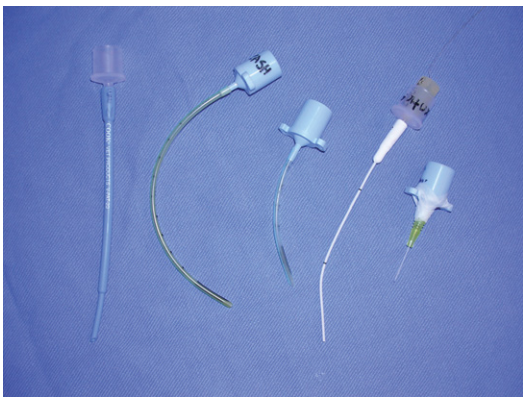


Figure 1.6 • Selection of endotracheal tubes that may be used with small exotic pets.





Figure 1.7 • (A) Various sizes of facemask are available. Clear masks allow better monitoring of patients during induction and anaesthesia; (B) a facemask can be adapted using a latex glove to create a smaller aperture for the patient's head.

Many animals, particularly mammals, will urinate and/or defecate during induction in chambers. Wetting of fur will increase the risk of hypothermia. The use of paper towels or incontinence pads to soak up fluids in the chamber will reduce fur contamination. The chamber should also be cleaned and disinfected between patients.

Facemasks should be close-fitting to reduce environmental contamination and resultant health risks to staff. Veterinary facemasks are usually cone-shaped to accommodate carnivore maxillae, but for species with shorter skulls, such as guinea pigs, human paediatric masks or those designed for cats may be more suitable. The masks should also be low volume, as a small increase in dead space may easily be the same as a small animal's tidal volume. For extremely small patients, such as rats, a syringe-case may be attached to the anaesthetic circuit to form a mask, or the end of the circuit used directly on the patient (see Fig. 4.8). Some anaesthetic circuits already have built-in masks (for example, rodent non-rebreathing circuit with nosecone, VetEquip®, Pleasanton, CA [see Fig. 4.5]), some may incorporate active gas-scavenging (for example, Fluovac®, International Market Supplies, Congleton, UK [Fig. 2.2]) (Hunter et al., 1984). Clear facemasks (Fig. 1.7) permit some visual assessment of the patient's head and are preferable to opaque masks.

As masks are usually plastic or rubber, they cannot be sterilised in an autoclave. They can be cleaned with most disinfectants or ethylene oxide sterilisation used if contamination with a particularly resistant infectious agent is suspected.

Some animals, for example birds, can be readily induced via facemask. For most species, however, induction is not as rapid and the restraint required can be stressful for the animal. Facemasks are most useful for maintaining anaesthesia in animals that cannot be intubated. The biggest disadvantage with a mask is a lack of airway control, and positive pressure ventilation (PPV) is not normally possible. (PPV may be performed in an emergency via a closely fitting facemask, but oesophageal inflation and gastric tympany may be produced.)

Endotracheal tubes for use in dogs and cats may be used in larger animals, but most exotic species require small uncuffed tubes. Many species have complete tracheal rings, laryngeal spasm may be a risk and narrow airways may easily be damaged by cuff over-inflation. For smaller patients, endotracheal tubes can be improvised from tubing available in the practice, for example, cut-off urinary catheters or intravenous catheters (with the stylet removed). If a large number of exotic pets are seen by the veterinary practice, it is worth investing in appropriate sized endotracheal tubes, from 1 to 5 mm diameter. A wide variety of types and sizes of endotracheal tubes are available (Fig. 1.6), some of which require the use of a stylet for placement.

Most new endotracheal tubes are excessively long, causing an increase in dead space, and should be shortened prior to use. To do this, the connector is removed from the tube and the tube cut to length before reattaching. (Do not cut the tip of the endotracheal tube, as this will leave a sharp end that may damage the patient's tracheal mucosa.) The aim is to place the tip of the tube within the animal's trachea above the bifurcation, with the connector



Figure 1.8 • Anaesthetic kit for exotic pets, including emergency drugs.



Figure 1.9 • Digital scales accurate to 1 g are vital for weighing small patients prior to calculating drug doses.

for the circuit at the lips to minimise dead space within the circuit. It is useful to have a selection of endotracheal tube sizes and lengths on hand, particularly for emergencies (Fig. 1.8). Always check that appropriate tubes are on hand before inducing anaesthesia.

Inspect endotracheal tubes before anaesthesia, particularly checking for lumen patency. It is easy for small tubes to become blocked with a small amount of respiratory secretion or other material. Tubes cannot be heat sterilised and are cleaned and sterilised as facemasks.

Many animals will breath-hold, or have reduced respiratory rate or tidal volume during anaesthesia. A *mechanical ventilator* is thus enormously useful in exotic-animal practice.

Prepare all equipment, including that required for anaesthesia and for the procedure to be performed, before inducing anaesthesia in the patient. This will minimise the anaesthetic time and thereby the risk to the animal.

Monitoring equipment

The most useful piece of anaesthetic monitoring equipment is a trained assistant. Assessment of physiological parameters is the cornerstone of patient monitoring. Other equipment may also be useful in different species, including bell or oesophageal stethoscopes, Doppler flow monitor, electrocardiogram (ECG) machine, capnograph and blood gas analysis.

Other equipment required

Scales used for cats are appropriate for medium-sized exotic pets, such as rabbits, but small kitchen-type digital scales (Fig. 1.9) that measure to the nearest gram are required for smaller animals. Most scales have a tare function, allowing the display to be tared after an empty container is placed on to the scales before the animal is weighed.

Supplemental heating is required for most exotic patients to maintain body temperature in patients both during and after anaesthesia. Equipment need not be as expensive as heated water or air blankets (Bair Hugger®, Arizant Healthcare, Eden Prairie, MN). Electric heat pads are useful, as are microwaveable heat pads and 'hot hands' (latex or nitrile gloves filled with warm water); most of these should be covered with a towel to prevent burning of the patient. It is important to warm fluids prior to administration to small patients that are more susceptible to hypothermia. Boluses of fluids in syringes may be warmed in a jug of warm water, while giving sets can be wrapped around 'hot hands' near the patient receiving a continuous rate infusion of fluids.

A *light source* is useful for intubation. For many species, an overhead directable theatre light or pen torch may be suitable. For other species with more caudal tracheal openings, a laryngoscope is advisable, for example with a Wisconsin size 0 or 1 blade. In some situations, an otoscope or small endoscope may be used. If the light source has been

in contact with an animal, it should be washed between patients to reduce the risk of cross-contamination.

Most other equipment is standard for veterinary practices, but smaller versions are required for smaller patients. For example, drug volumes are more likely to necessitate the use of 1 ml syringes and 25 gauge needles, and insulin syringes are especially useful when drug dilutions are to be performed for very small animals. Small over-the-needle catheters are useful for many procedures, including intravenous fluid or drug administration and as endotracheal tubes in tiny patients. Giving sets used in larger animals may not be readily calibrated to provide small volumes of fluids. The use of infusion pumps, burette giving sets or syringe-driver infusion pumps is extremely useful where continuous rate infusions are required. In many patients, fluids are administered as boluses. Although proprietary small-gauge intraosseous needles are available, hypodermic needles can be used as intraosseous catheters in small patients (see Fig. 4.3).

EQUIPMENT PREPARATION

Before using an anaesthetic system on a patient some routine checks should be performed. These include checking that all connections are secure and that sufficient gases (for example, oxygen) and volatile agents are available. The anaesthetic circuit should be leak-tested, by closing the expiratory end (most have valves that can be closed), placing a thumb over the end that connects to the patient and filling the circuit with oxygen. Endotracheal tubes should be checked for patency and cuffs (if present) inflated to check for leaks. Anaesthetic time can be greatly minimised by collecting all equipment required for anaesthesia and the procedure to be performed, before the patient is induced.

At the end of anaesthesia, endotracheal tubes, facemasks and anaesthetic circuits should be cleaned between patients. Sterilisation is also necessary in some instances, particularly with endotracheal tubes. The anaesthetic machine oxygen should be switched off and the vaporiser re-filled with volatile agent.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

All animals should be assessed before anaesthesia, including a detailed history and clinical examination (including an accurate body weight). Further investigations may be indicated depending on the animal's condition. This assessment will allow the clinician to gauge the anaesthetic risks and to select an appropriate protocol.

Weigh animals accurately, particularly before administration of injectable drugs. Digital scales with 1 g increments are necessary for small species (Fig. 1.9).

> Clinical assessment may identify signs of illness which require attention before anaesthesia is performed, or factors that will adversely affect anaesthesia.

Many pet mammals are obese. This may compromise cardiopulmonary function during anaesthesia by reducing cardiac reserve (Carroll et al., 1999), causing hypoventilation (Ahmed et al., 1997).

Exotic pets are often dehydrated or otherwise debilitated when presented to the veterinary clinician. In many cases it is advisable to postpone anaesthesia while correcting fluid deficits and/or administering nutritional support. For some patients, provision of appropriate diet and environmental conditions may be sufficient for the patient to ingest food and water. Unfortunately, many are beyond this stage and require intervention. Nutritional support may involve hand-feeding or assist-feeding. The oral route is useful for administration of maintenance fluids or in those animals with mild dehydration. Subcutaneous fluids are useful in many species, but absorption may be slow, particularly in hypothermic animals. Intraperitoneal fluids are rapidly absorbed, but administration carries the risk of visceral puncture. Intravenous or intraosseous fluids are excellent methods of accessing the circulatory system for replacement of moderate to severe fluid deficits, but are obviously more technically demanding to place than other techniques.

The choice of anaesthetic protocol will be based on findings at this stage. An appreciation of the patient's current health status, along with the purpose of the anaesthesia, will allow the clinician to select the most appropriate drugs. A debilitated animal will likely be unable to metabolise drugs well, and a prolonged recovery may reduce chances of survival. If surgery is indicated, analgesia should be included in the anaesthetic protocol, perhaps synergistically with other agents.

ANAESTHETIC DRUGS

Most anaesthetic agents are not licensed for use in exotic pets. Some drugs, for example narcotic analgesics, may be controlled under national legislation. These may require specific storage facilities and/or records of their purchase and use. It is good practice to keep any drugs with the potential for human abuse in a locked cupboard.

The doses for most agents have not been experimentally elucidated for exotic species. Differences in physiology and metabolism between species will alter the effects of drugs, including safety margins. Doses relevant for larger animals, such as dogs, will rarely be transferable to small species, such as rodents, with high metabolic rates. Other species, such as reptiles, have extremely slow metabolic rates.

There are several classes of drugs that produce anaesthesia and effects seen may differ between species (and often also between individuals within a species). Although there is a temptation to use a single agent in order to simplify the anaesthetic protocol, the use of multiple agents from different classes allows the clinician to obtain a more balanced anaesthesia, for example including analgesia if required. If multiple drugs are used, doses of individual drugs can be lowered, reducing their side effects (except where two agents have the same side effects, in which case they may be additive).

Besides a lack of licensed drugs that have been rigorously tested, other difficulties encountered in using anaesthetic

drugs in exotic pets include technical problems associated with drug administration, and difficulties with anaesthetic monitoring of animals that are often much smaller or have different anatomy and physiology than more common pet species. In preparing an anaesthetic protocol, consideration should be given to the patient's health and the procedure to be performed during anaesthesia. For example, phlebotomy may require sedation or a brief anaesthesia only, whilst surgery will necessitate a deeper plane of anaesthesia for a more prolonged period, as well as appropriate analgesia. Many anaesthetic problems are associated with the postoperative period and peri-anaesthetic management is vital for a successful outcome.

The ensuing chapters aim to discuss species differences affecting anaesthesia, but the following section discusses anaesthetic agents in general.

Mechanisms of action

General anaesthetics affect the central nervous system; predominantly the higher functions. Respiratory control is often impaired during general anaesthesia, as is temperature homeostasis.

Many anaesthetic agents inhibit nicotinic acetylcholine receptors, in particular the volatile agents and ketamine (Tassonyi et al., 2002). Modulation of these receptors is not directly involved in the hypnotic component of anaesthesia, but may contribute to analgesia with some agents.

Local anaesthetics

These drugs are weak bases and block sodium ion channels, and thence stop both motor and sensory nerve transmission (Skarda, 1996). Local anaesthetics may be used to provide analgesia locally, and to reduce the doses of sedatives and general anaesthetics required (Hedenqvist and Hellebrekers, 2003). The use of regional anaesthesia (as opposed to general anaesthesia) has been shown to allow earlier rehabilitation and shorten hospital stays in patients (Capdevila et al., 1999).

Local anaesthetics can be administered by several routes, including topical sprays, liquids or creams, or by local infiltration, intrapleurally and epidurally. The most commonly used topical agent is EMLA cream (AstraZeneca, Södertälje, Sweden), which contains lidocaine (lignocaine) and prilocaine; it produces full-skin-thickness anaesthesia within 60 min of application (Nolan, 2000). Topical application of liquid local anaesthetics, such as proxymetacaine, will result in corneal and conjunctival anaesthesia. Lidocaine (lignocaine) is commonly sprayed on to the larynx of animals prone to laryngeal spasm prior to intubation. Local anaesthetics can be infiltrated into skin and underlying tissues to assist minor procedures, but a sedative or light plane of anaesthesia is often required to immobilise the patient concurrently. In larger animals, certain anatomical sites have a well-defined nerve supply, and individual nerves can be anaesthetised (for example the paravertebral nerve block).

Local anaesthetics administered into the epidural space between the dura mater and the wall of the vertebral canal will cause both motor and sensory nerve blockade. Other agents, such as opioids, ketamine or xylazine, are commonly used with local anaesthetics in epidurals for analgesia or anaesthesia (Nolan, 2000). If opioids are administered without local anaesthetics, sensory block only will be produced.

Lipid solubility affects the duration of action, with bupivacaine being more lipid and, therefore, having a longer duration than lidocaine (lignocaine). The duration of action of lidocaine (lignocaine) is 60–90 min, and is increased by adding adrenaline (epinephrine). Bupivacaine has a high rate of protein binding, which prevents absorption, and the duration is 2–6 h (Hedenqvist and Hellebrekers, 2003). Bupivacaine has been shown to be myotoxic in rabbit extraocular muscles (Park and Oh, 2004). Ropivacaine is similar to bupivacaine, but is less cardiotoxic. All three drugs undergo hepatic metabolism by cytochrome P-450.

A major cause of anaesthetic mortality is human error leading to anaesthetic overdosage and to hypoxia (Jones, 2001). Overdoses of local anaesthetics result in systemic toxicity, which causes hypotension, ventricular arrhythmia, myocardial depression and convulsions. The maximum safe doses for most species are 4 mg/kg for lidocaine (lignocaine) and 1–2 mg/kg for bupivacaine (Dobromylskyj et al., 2000).

MS-222 (tricaine methane sulfonate) is a soluble local anaesthetic. It is commonly used to anaesthetise fish and amphibian species (Bowser, 2001).

Pre-anaesthetic medication

Drugs may be administered before anaesthetic induction for several reasons. These include sedation to: reduce the stress of anaesthetic induction (to handlers or patients), reduce the dose of other agents required, reduce the risk of side effects that may occur with anaesthetic agents used or surgery performed, or smooth anaesthetic induction and recovery. For most exotic pet species, long-acting pre-medications are not used, as rapid recovery after anaesthesia is desirable. It is, therefore, also preferable to use inhalation rather than injectable anaesthetic agents where possible to provide a speedier recovery.

BOX 1.1 Groups of sedative and anaesthetic agents

- Alkyl phenol, e.g. propofol
- Alpha-2-agonists, e.g. medetomidine
- Benzodiazepines, e.g. midazolam
- Butyrophenones, e.g. fluanisone
- Dissociative agents, e.g. ketamine
- Local anaesthetics, e.g. lidocaine
- Opioids (narcotic analgesics), e.g. fentanyl
- Phenothiazine derivatives, e.g. acepromazine
- Steroid agents, e.g. alfaxalone
- Volatile agents, e.g. isoflurane

A simple form of pre-anaesthetic medication is to use local anaesthetic ointment to anaesthetise the skin before intravenous access is used to induce anaesthesia (Flecknell et al., 1990). Where pre-anaesthetic medication is given to produce sedation, the animal is left in a quiet area for 10–30 min after administration to allow the drug to take effect (Hedenqvist and Hellebrekers, 2003).

Anticholinergic drugs reduce bronchial and salivary secretions. This is desirable because these secretions may be problematic in small animals, causing airway occlusion. In some species, salivary secretions may become more viscous after anticholinergics (Flecknell, 1996). Atropine can be used to protect the heart from vagal inhibition, or to treat bradycardia caused by opioids. Care should be taken in species with normally high heart rates, such as birds. An overdose of anticholinergic agents may cause seizures (Hedenqvist and Hellebrekers, 2003). If administered prior to alpha-2-agonists, anticholinergics may initially prevent bradycardia. However, the initial hypertension associated with the alpha-2-agonist may be potentiated.

Atropine is used in preference for cardiac emergencies as it is faster in onset and shorter in duration than glycopyrrolate. The latter drug has a more selective anti-secretory action, and does not cross the blood–brain barrier or placenta, therefore, causing minimal central nervous system (CNS) and fetal effects (Flecknell et al., 1990; Heard, 1993). Glycopyrrolate is used in preference in rabbits and rats, which destroy atropine with hepatic atropinesterase (Harkness and Wagner, 1989; Olson et al., 1993).

Diazepam, midazolam and zolazepam are *benzodiazepines*. These drugs are weak bases, and act by potentiation of gamma-aminobutyric acid (GABA). They produce sedation and good skeletal muscle relaxation and are anticonvulsant (Brunson, 1997). These agents cause minimal cardio-respiratory depression (Short, 1987), but also do not provide analgesia (Hedenqvist and Hellebrekers, 2003). Hyperalgesia may occur, and analgesia should be provided if surgery has been performed (Flecknell, 1996). Flumazenil is a specific antagonist to the benzodiazepines (Amrein and Hetzel, 1990; Pieri et al., 1981). Some reports have shown diazepam to have toxic effects on liver cells (Strombeck and Guildford, 1991). Diazepam usually comes as a propylene glycol formulation that must be administered intravenously, and cannot be mixed with other agents. Although midazolam is shorter acting, it is more potent and is water-soluble. It can be mixed with other agents, such as atropine, fentanyl, Hypnorm® (Janssen Pharmaceuticals, Beerse, Belgium) and ketamine. Zolazepam is potent and long acting (Heard, 1993).

Opioids are often administered with benzodiazepines, to increase the sedation produced. The benzodiazepines are also frequently used to potentiate dissociative anaesthetics and to improve muscle relaxation (Heard, 1993). Diazepam or midazolam is often combined with ketamine. Zolazepam is prepared in combination with the dissociative agent tiletamine (as Zoletil®, Virbac, Peakhurst, NSW; Telazol®, Fort Dodge, IA). This drug may cause nephrotoxicity in rabbits (Hedenqvist and Hellebrekers, 2003).

Phenothiazine derivatives, such as acepromazine, are tranquillisers, which produce sedation by blocking dopamine centrally. Peripheral alpha-adrenergic antagonistic effects are also seen (Brunson, 1997). No analgesia is produced. These agents reduce the dose of other agents required to produce surgical anaesthesia, including anaesthetics, hypnotics and narcotic analgesics. Disadvantages include a long duration of action, variable response, moderate hypotension due to peripheral vasodilation, depressed thermoregulation and a lowered CNS seizure threshold (Hedenqvist and Hellebrekers, 2003; Short, 1987). These agents should be avoided in dehydrated patients (Flecknell, 1996).

The *butyrophenones* include droperidol, fluanisone and azaperone. These act similarly to the phenothiazines (Brunson, 1997), but produce less severe hypotension. They are often used in neuroleptanalgesic combinations, for example droperidol with fentanyl (Innovar-Vet®, Janssen Pharmaceuticals, Ontario, Canada) or fluanisone with fentanyl (Hypnorm®, Janssen Pharmaceuticals, Beerse, Belgium) (Flecknell, 1996). Hypnorm® is commonly used in combination with midazolam to produce surgical anesthesia, for example in rabbits or rodents (Hedenqvist and Hellebrekers, 2003). Azaperone is used in pigs, causing immobilisation with minimal side effects (Swindle, 1998). Anticholinergic agents are used to avoid some of the adverse effects seen, which may include bradycardia, hypotension, respiratory depression, hypoxia, hypercapnia and acidosis. The butyrophenones have a long duration of activity, and may produce paradoxic excitement and aggression in some animals (Heard, 1993).

The *alpha-2-adrenergic agonists* medetomidine and xylazine are potent sedatives, also causing muscle relaxation, anxiolysis, and variable analgesia. Action at the alpha-2-adrenoceptors inhibits presynaptic calcium influx and neurotransmitter release (Hedenqvist and Hellebrekers, 2003). These agents potentiate most anaesthetic drugs.

Cardio-respiratory depression with these agents varies between dose, species and other agents (Short, 1987). Respiratory depression is observed in most species and cardiac effects, such as bradycardia, bradyarrhythmias and hypotension, vary between species and dose. Initially hypertension is seen, followed by slight hypotension, bradycardia and reduced cardiac output (Hedenqvist and Hellebrekers, 2003). These agents depress insulin release and thence cause hyperglycaemia (Feldberg and Symonds, 1980; Lukasik, 1999). Diuresis is due to a decrease in antidiuretic hormone and a direct renal tubular effect (Greene and Thurmon, 1988).

Xylazine is a mixed alpha-2/alpha-1-agonist (Lukasik, 1999), and may cause cardiac arrhythmias in some species (Flecknell, 1996). As xylazine increases uterine tone in some species, it should be avoided in pregnant animals (Hedenqvist and Hellebrekers, 2003). Xylazine is not very effective as a sole agent in most exotic species, but may be used in combinations (Heard, 1993). Medetomidine is more selective for alpha-2 adrenoceptors (Brunson, 1997), is more potent and reportedly has fewer side effects than xylazine (Virtanen, 1989). The effects of these drugs vary between species; for example, the analgesic

properties of medetomidine are weak in rabbits, guinea pigs and hamsters.

These agents are most commonly used in combination with ketamine, which will offset the bradycardia and result in hypertension (Lukasik, 1999). Combinations with opioids or benzodiazepines will enhance sedation and analgesia (Hedenqvist and Hellebrekers, 2003).

A major advantage with alpha-2-adrenergic antagonists is that they can be reversed, but administration of the antagonist should be delayed for 45–60 min if ketamine has been given, as ketamine alone causes tremors and muscular rigidity (Frey et al., 1996). Atipamezole is more short acting than medetomidine and is usually not administered for 30–40 min after medetomidine to avoid resedation (Harcourt-Brown, 2002). If resedation occurs, the atipamezole may be repeated.

Atipamezole is a specific antagonist for medetomidine, but will also partially reverse xylazine (Flecknell, 1996). Yohimbine is a more specific antagonist for xylazine (Hedenqvist and Hellebrekers, 2003). Intravenous administration of these antagonists is not recommended.

Many *narcotic analgesics* are used to cause moderate sedation where analgesia is also required. They also reduce the doses of anaesthetic drugs necessary to produce anaesthesia. They are often combined with neuroleptics (tranquillisers or sedatives). Drugs include morphine, pethidine, buprenorphine, butorphanol and fentanyl. Respiratory depression is the most common side effect; some will also affect gastrointestinal motility (Flecknell, 1996).

Inhalation anaesthesia

Gaseous anaesthetic agents used in exotic pets are predominantly halogenated hydrocarbons, halothane or halogenated ethers, such as isoflurane and sevoflurane. These agents interact with receptors in the CNS, enhancing the inhibitory neurotransmitters GABA and glycine (Hedenqvist and Hellebrekers, 2003; Mihic et al., 1997). In most exotic pet species, various gaseous anaesthetic agents can be used to induce and/or maintain anaesthesia. These agents are ideal for lengthy procedures, as the recovery period is not prolonged with longer administration of agents (unlike many injectable agents). It is vital to check equipment prior to anaesthesia, ensuring that it is functional and that sufficient gases and anaesthetic agents are available close at hand.

Isoflurane is the most commonly used agent, but sevoflurane can be used for most species. These agents are volatile liquids at room temperature and vaporisers are used to add them to inspired gases, usually mixed with oxygen. After inspiration, the agent diffuses down concentration gradients, passing from airways to the blood and thence to tissues including the brain.

The minimum alveolar concentration (MAC) is a measure used to define the potency of a volatile anaesthetic agent. It is the concentration of gaseous anaesthetic agent required to prevent movement in 50% of patients in response to a noxious stimulus (Eger et al., 1965), and is similar for animals of the same species, but may differ slightly between species. MAC values are end-tidal concentrations of anaesthetic, rather than vaporiser settings. Values will vary slightly between studies if different 'noxious stimuli' are used. MAC values are lower after certain pre-medication drugs have been administered (Turner et al., 2006). The values also decrease with age, and higher concentrations of agent are required to anaesthetise neonates (Hedenqvist and Hellebrekers, 2003).

The MAC value is inversely related to potency; hence agents with low MAC values will be more potent and require low inspired concentrations to produce a particular effect. Agents with a high lipid-gas partition coefficient (λ) will have a low MAC; the converse is also true. For example, halothane's blood-gas λ is 2.5 and MAC (in the dog) is 0.87, isoflurane's λ is 1.4 and MAC (dog) is 1.28, and λ for nitrous oxide is 0.5 while MAC (dog) is 222 (Steffey, 1994). MAC is fairly constant between species (Table 1.1), varying by less than 20% between species (Ludders, 1999). For example, MAC for halothane is 0.87% in dogs and 0.95% in rats; MAC for isoflurane is 1.28% in dogs and 1.38% in rats (Flecknell, 1996; Steffey, 1994).

Another important factor for volatile agents is the equilibration time, the time taken for the drug to act. *Blood solubility* affects the time until the anaesthetic agent reaches the brain and spinal cord, and the effects of anaesthesia are seen. Isoflurane produces more rapid induction, as it is less soluble in blood than halothane (Hedenqvist and Hellebrekers, 2003). Agents that are relatively insoluble in blood (with a low blood-air λ) will diffuse rapidly from the circulation into the airways and be expired, causing a rapid recovery from anaesthesia. Halothane has a relatively high blood-air λ, and is lost slowly into the airways; ventilation rate, thus, limits the expiration of and recovery from this agent. An agent's *lipid solubility* also affects potency, with highly lipid-soluble agents being

Table 1.1: Minimum alveolar concentrations (MAC , %) for volatile anaesthetic agents in selected species

ANAESTHETIC	DOG	MOUSE	PIG	PRIMATE	RABBIT	RAT
Halothane	0.87	0.95	1.25	1.15	1.39	0.95
Isoflurane	1.28	1.41	1.45	1.28	2.05	1.38
Nitrous oxide	222	275	277	200	–	150

(Drummond, 1985; Flecknell, 1996; Mazze et al., 1985; Steffey, 1994; Valverde et al., 2003)

more potent. Similarly, these agents will accumulate in adipose tissue and recovery from anaesthesia may be slow.

Most gaseous anaesthetic agents induce anaesthesia rapidly, do not require metabolism to any great degree, and allow rapid recovery when the agent is no longer administered to the patient. They are thus considered relatively 'safe' anaesthetics. Cardio-respiratory and renal blood flow depressions are dose-dependent (Steffey, 1996). Disadvantages include the smell and airway irritation, which may lead to breath holding in some species, such as rabbits and reptiles, and poor analgesia. A pre-medicant may be used to sedate the animal and reduce the former disadvantage prior to gaseous induction. An alternative is to induce the animal with injectable agents and maintain anaesthesia using a volatile agent.

Inhalation agents do necessitate the purchase of anaesthetic machines and circuits. While this is not absolutely necessary for anaesthesia with injectable agents, it is advisable to use an anaesthetic machine during all anaesthetic procedures, as oxygen supplementation should always be administered. This is particularly important when using injectable agents (see below) that may compromise cardio-respiratory function.

Waste gases may contaminate the environment and be hazardous to humans, particularly with halothane that is metabolised more than isoflurane. Excess gas should, therefore, be scavenged effectively (Hedenqvist and Hellebrekers, 2003). It is good practice to monitor environmental concentrations of inhalational agents, to assess scavenging techniques and possible health risks for staff.

Halothane

This agent is derived from chloroform, is unstable in light and very soluble in rubber. Halothane has a high lipid solubility and low MAC; these result in a potent anaesthetic with rapid induction. However, muscle relaxation is limited and analgesia minimal. Recovery may be delayed after prolonged, deep anaesthesia (Flecknell, 1996).

Several cardio-respiratory changes are seen with halothane use. Moderate respiratory depression occurs due to a dose-dependent decrease in medullary carbon dioxide sensitivity. Myocardial contractility is reduced, sympathetic ganglion blockade leads to bradycardia and relaxation of vascular smooth muscle reduces diastolic blood pressure. The myocardium is also sensitised to catecholamines, with the risk of arrhythmias (Brunson, 1997).

Twenty per cent of absorbed halothane gas undergoes hepatic metabolism. Hepatic enzymes are, therefore, induced during halothane anaesthesia. If hypoxia is present, hepatic metabolism may produce radicals, which may lead to hepatotoxicity (Ludders, 1999). Risks to veterinary staff include hepatotoxicity, and it may be teratogenic in women. Good scavenging is required to reduce environmental contamination.

Halogenated ethers

These include isoflurane, sevoflurane and desflurane. If overdosed, these agents tend to cause apnoea before cardiac arrest. This allows the anaesthetist to counter the adverse effects and provide respiratory support, and to avoid cardiac problems.

Isoflurane gas is non-irritant (Flecknell, 1996). The MAC for isoflurane is similar to halothane, but the blood-air λ is less, producing more rapid induction and recovery than halothane. Moderate analgesia and muscle relaxation are produced.

Although respiratory depression is similar to that seen with halothane, cardiac effects are much less pronounced. Vasodilatory effects are seen, for example in the coronary vessels (Brunson, 1997). Heart rate and arterial blood pressure are not significantly affected, and the myocardium does not become sensitised to catecholamines (Hedenqvist and Hellebrekers, 2003). Studies in rabbits have shown that isoflurane produces reactive oxygen species that contribute to protection against myocardial infarction (Chiari et al., 2005; Tanaka et al., 2002; Tessier-Vetzel et al., 2005). Very little absorbed isoflurane is metabolised (Eger, 1981), with most being expired. Only 0.2% is metabolised in the liver; this makes it a safer anaesthetic in animals with reduced hepatic metabolism. Induction and recovery are rapid with isoflurane, and it is routinely used in veterinary practices for anaesthesia of all exotic pet species.

Sevoflurane and desflurane are similar to isoflurane. *Sevoflurane* has negligible airway irritant effects (Patel and Goa, 1996), and, therefore, is less stressful for animals induced in a chamber or via facemask. This agent has a very low solubility in blood and, therefore, induction and recovery are more rapid than with isoflurane (Hedenqvist and Hellebrekers, 2003). Sevoflurane is also protective against myocardial infarction (Chiari et al., 2004). This agent is metabolised in a similar manner to isoflurane. However, it is unstable in soda lime, forming haloalkenes that may be nephrotoxic in certain species. Antioxidant supplementation with vitamin E and selenium has been shown to protect against damage to DNA caused by repeated sevoflurane anaesthesia (Kaymak et al., 2004).

Desflurane undergoes the least metabolism of the volatile agents (Koblin, 1992), and induction and recovery are the most rapid (Eger, 1992). Toxicity is very low with this agent (Hedenqvist and Hellebrekers, 2003).

Nitrous oxide

Although this agent has a place in anaesthesia, its extremely low potency (with high MAC) in animals minimises its usefulness. Solubility in blood, oil and fat is poor, and, therefore, uptake and equilibration are rapid (Hedenqvist and Hellebrekers, 2003). Cardio-respiratory effects are minimal and excellent analgesia is produced. The second gas effect means that nitrous oxide may be useful in conjunction with another volatile agent to increase the rate of induction. At least 33% oxygen should always be administered with nitrous oxide, in order to avoid hypoxia in the patient (Ludders, 1999). It is more usual to have a 50:50 or 60:40 mix of nitrous oxide to oxygen.

During recovery, nitrous oxide diffuses into the airways from the blood, reducing the volume of inspired air and associated oxygen intake; higher flow rates and/or oxygen

are, therefore, necessary during recovery to prevent diffusion hypoxia. Nitrous oxide is not absorbed by either soda lime or activated charcoal. This gas should not be used, therefore, in a closed anaesthetic circuit and there should be active scavenging to the building's ventilation outlet. Nitrous oxide may diffuse into gas-filled intestines and is, therefore, not recommended in herbivorous species (Hedenqvist and Hellebrekers, 2003). Chronic exposure to nitrous oxide may increase rates of abortion and teratogenicity in veterinary staff.

Injectable anaesthetic agents

Routes of administration for these agents are intravenous, intramuscular, subcutaneous and intraperitoneal. Many drugs may be irritant; care should be taken to calculate and measure doses accurately, ensure volumes administered are not excessive for the size of patient (particularly for intramuscular injections), and administer drugs using an appropriate technique. Another problem that occurs when using injectable agents is inter- and intra-species variation in response to the drugs. It is not always possible to obtain a reported drug dose, and extrapolations may need to be drawn from similar species. Individual animal variation is often dependent on current disease processes, and pre-anaesthetic assessments are vital in identification of any factors that may adversely affect the patient during anaesthesia.

Intravenous induction of anaesthesia is usually the most rapid and many agents are titratable. However, intravenous access is technically difficult in many exotic pet species or may only be possible in sedated animals. The approach to anaesthesia may, therefore, be different to other species.

The possibility of 'topping up' anaesthetic agents may arise during the use of injectable agents. It is advisable to administer further doses by the intravenous route, so that the dose may be easily titrated to effect. To obtain accuracy of dose delivery, infusion pumps or syringe drivers should be used. Problems may arise if redistribution of the drug occurs, such as with barbiturates, and recovery may be prolonged. With some agents, such as alfaxalone/alphadolone, recovery is rapid (Cookson and Mills, 1983), and repeat doses or a continuous rate infusion may be used for prolonged anaesthesia. Similarly, propofol has little cumulative effects and may be used as the sole anaesthetic agent (Aeschbacher and Webb, 1993; Blake et al., 1988; Brammer et al., 1993). Opioids may also be added to a mix of agents for total intravenous anaesthesia (TIVA). If benzodiazepines are used concomitantly with an opioid, relative overdose of the benzodiazepine may occur due to its longer duration of action and it is preferable merely to top up the opioid component. Ketamine is sometimes used to prolong anaesthesia, but incremental doses prolong recovery and severe respiratory depression may occur (Flecknell, 1996).

Propofol is an alkyl phenol (Glen, 1980; Glen and Hunter, 1984) with poor water solubility. It is administered intravenously and produces anaesthesia in many species by enhancing GABA-receptor function (Hedenqvist and Hellebrekers, 2003). Perivascular administration is not irritant (Morgan and Legge, 1989), but intramuscular administrations will only cause sedation. Induction of anaesthesia is usually rapid (Edling, 2006). Propofol is redistributed rapidly, tissue accumulation is minimal and propofol is rapidly metabolised in the liver, resulting in rapid recovery (Stoelting, 1987). Propofol has been shown to have anti-oxidant effects (Mathy-Hartert et al., 1998; Murphy et al., 1993) and attenuated endotoxin-induced acute lung injury in rabbits (Kwak et al., 2004).

Propofol reduces both carotid body chemosensitivity (Jonsson et al., 2005) and baroreceptor responsiveness (Memtsoudis et al., 2005). Side effects include a moderate fall in systolic blood pressure, a small reduction in cardiac output (Sebel and Lowdon, 1989), and significant respiratory depression (Glen, 1980). The respiratory depression may result in a reduced respiratory rate or reduced tidal volume (Watkins et al., 1988), and oxygen should be supplemented. The cardio-respiratory depression is dose-dependent (Machine and Caulkert, 1996). Slow administration will avoid apnoea (Hedenqvist and Hellebrekers, 2003), which is common in rabbits. Cerebral blood flow and oxygen consumption are reduced, and intracranial pressure lowered by propofol. In pigs, myocardial contractility is reduced. Hepatic, renal, platelet and coagulation functions are not affected by propofol (Sear et al., 1985). Analgesic properties are minimal and doses required for analgesia are associated with hypotension, and reduced heart rate and arterial blood pressure. Premedication with a number of agents will reduce the dose of propofol required for anaesthesia (Hellebrekers et al., 1997).

Barbiturates are infrequently used to produce anaesthesia in exotic pets as their therapeutic index is low and effects irreversible. Most are highly alkaline and irritant to tissues, excepting pentobarbital that has a relatively neutral pH. Cardio-respiratory depression is produced, which is dose-dependent. Analgesia is poor with these agents, and hyperalgesia may be produced (Heard, 1993).

Steroid anaesthetic agents

Alfaxalone and *alphadolone* are both steroids, with a wide safety margin (Child et al., 1971; Child et al., 1972b; Child et al., 1972c). The usual route of administration is intravenous. Intramuscular or intraperitoneal injection is non-irritant, and will also produce effects, but these are variable (Green et al., 1978). Intravenous injection causes smooth induction of anaesthesia with rapid recovery. Moderate hypotension may be seen (Child et al., 1972a; Dyson et al., 1987). Continuous rate infusions or boluses have been used in various species to maintain more prolonged anaesthesia (Flecknell, 1996).

Dissociative anaesthetic agents

Ketamine and tiletamine are lipophilic cyclohexamines, with antagonistic effects at *N*-methyl-D-aspartate (NMDA) receptors. The resulting depression of cortical associative areas produces a 'dissociative state' (Hedenqvist and Hellebrekers, 2003). Moderate respiratory depression occurs, but bronchodilation is also present. The gag reflex

is retained, but may not prevent aspiration if regurgitation or vomition occurs (Heard, 1993). The corneal reflex is lost in many species and ocular lubricants should be applied to prevent damage to the corneas or spectacles. An increase in skeletal muscle tone is produced and purposeful muscle movements may occur during anaesthesia. Although myocardial depression occurs, an increase in blood pressure is seen due to sympathetic nervous system stimulation. Analgesia with these agents is dose-dependent. The drugs are metabolised in the liver.

Ketamine can be administered intramuscularly, intravenously or intraperitoneally to produce sedation with apparent lack of awareness (White et al., 1982). The high doses required in rodents to produce surgical anaesthesia can be associated with severe respiratory depression (Green, 1981). Laryngeal and pharyngeal reflexes are usually retained, but an increase in salivary secretions may cause airway obstruction. Anticholinergics may be used to reduce these bronchial and salivary secretions (Flecknell, 1996).

Ketamine is extremely useful in primates. In many species, combining ketamine with alpha-2 antagonists, benzodiazepines or phenothiazines produces anaesthesia. Ketamine administered chronically will induce hepatic enzymes, and subsequent doses may be less effective (Marietta et al., 1975). Recovery may also be prolonged after ketamine, and hallucinations and mood alterations may occur (Wright, 1982).

It has a low pH, and may cause discomfort on injection (Heard, 1993). There are several reports of acute muscle irritation and chronic myositis following injection with ketamine and xylazine (Beyers et al., 1991; Gaertner et al., 1987; Latt and Echobichon, 1984; Smiler et al., 1990). Discomfort may cause the animal to self-traumatise the body part after recovery.

Tiletamine is two to three times as potent as ketamine, and has a longer duration (Short, 1987). Nephrotoxicity to high-dose tiletamine/zolazepam has been reported in New Zealand white rabbits (Brammer et al., 1991).

Neuroleptanalgesic combinations

These combinations are useful where analgesia is required along with anaesthesia. These combinations include an opioid that is a narcotic analgesic, and a tranquilliser or sedative (the neuroleptic) that suppresses some of the opioid's side effects. Disadvantages of these combinations include a moderate to severe respiratory depression, poor muscle relaxation, along with hypotension and bradycardia in some cases (Flecknell, 1996). Assisted ventilation is not always required, but is beneficial in reducing hypercapnia and acidosis during prolonged anaesthetics. The biggest advantage of these combinations is the reversibility of the opioid by opioid-antagonists, such as naloxone, mixed agonist/antagonists, such as nalbuphine, or partial agonists, such as buprenorphine or butorphanol (Flecknell et al., 1989).

Used alone, muscle relaxation is poor with opioids; this can be improved by adding a butyrophenone. Common combinations are fentanyl and fluanisone (Hypnorm®, Janssen, Janssen Pharmaceuticals, Beerse, Belgium), and fentanyl and droperidol (Innovar-Vet®, Janssen, Pharmaceuticals, Ontario, Canada). The former combination produces good surgical anaesthesia when a benzodiazepine, such as midazolam or diazepam, is also administered. The latter neuroleptanalgesic combination produces less predictable anaesthesia (Flecknell, 1996; Marini et al., 1993).

Opioids, such as fentanyl or alfentanil, may also be used in combination with benzodiazepines. The opioids provide potent analgesia and are often included in anaesthetic combinations for this reason. High doses of opioid will cause respiratory depression, but this can be managed using intermittent positive pressure ventilation in intubated anaesthetised patients (Flecknell, 1996).

PERI-ANAESTHETIC SUPPORTIVE CARE, INCLUDING ANALGESIA

Supplemental *heating* will be necessary in almost all exotic pets. Larger species, such as minipigs, may not require warming if anaesthetised in a veterinary practice, but are likely to if anaesthetised outdoors or in an unheated house. Insulation of the animal, for example using bubble-wrap, to prevent heat loss may be sufficient to maintain body temperature. In most small patients, however, additional heating should be provided, such as overhead heat lamps, warm-air blankets (for example Bair Hugger®, Arizant Healthcare, Eden Prairie, MN), electric heat mats or hot water bottles. Care should be taken not to overheat patients, and mats and bottles are usually covered with a layer of towelling to prevent contact burns. Thermostatically controlled heating blankets are available (for example Homeothermic Blanket System®, International Market Supply Ltd, Cheshire, UK).

During anaesthesia, the patient's *position* should be monitored. The exact positioning will depend on the procedure to be performed, but the head and neck should be extended to prevent the tongue or soft palate from obstructing the larynx. In general, the head and thorax should be maintained slightly higher than the abdomen to avoid abdominal viscera compressing the lungs. Respiratory movements should not be impeded; in avian species, for example, positioning should allow keel movement. If the patient is intubated, the endotracheal tube should be attached to the animal using either bandage material or adhesive tape (for example, Micropore®, 3M, St Paul, MN). It is also usually helpful to attach the anaesthetic circuit to the surface on which the animal is positioned, as the weight of the circuit may pull on the endotracheal tube and/or the patient. If a change in patient position is required, for example during radiography, it is often simpler temporarily to disconnect the patient from the circuit while moving the animal (Flecknell, 1996).

O*cular lubricants* should be used in most animals to prevent desiccation and trauma to the corneas (or spectacles in snakes and lizards) during anaesthesia and recovery. It may be possible to tape the eyelids closed (for example, using Micropore® tape, 3M, St Paul, MN).

O*xygen* therapy is most easily, and least stressfully, provided in a chamber before and after anaesthesia. If an oxygen

chamber is not available, use of an anaesthetic circuit carrying 100% oxygen into a small kennel or carry box will increase the inspired concentration of oxygen for the animal. This can be useful both before anaesthesia and during recovery, particularly for mammalian and avian species. (Provision of high concentrations of inspired oxygen is often contraindicated in reptiles, as it will depress their respiratory drive.) If high flow rates are being used, ensure the gas flow does not lower the animal's environmental temperature.

Fluids may be required to stabilise the debilitated patient before anaesthesia. They also assist when anaesthetic agents depress cardiovascular function during anaesthesia, or in maintaining circulation and metabolism of anaesthetic drugs. In cases of fluid loss intra-operatively, such as haemorrhage, administration of parenteral fluids may well be life saving. Fluids can be administered up to rates equivalent to 10% of circulating volume per hour (Flecknell, 1996).

In most patients, fluid can be administered at 10 ml/kg/h using Hartmann's solution or 0.9% saline (Flecknell, 1996). Most animals can cope with the loss of up to 10% of their circulating volume acutely, but clinical signs of hypovolaemia and shock will be seen if >15–20% is lost. Whole blood transfusions are likely to be required if >20–25% of the circulating blood volume is lost. Blood transfusions have been performed in many species, with preference given to a donor animal of the same species as the recipient. If whole blood is not available, colloids can be given to expand circulatory volume; if neither blood nor colloids are available, Hartmann's solution or 0.9% saline may be administered, although crystalloids will redistribute rapidly throughout the body. If intravenous access is not possible, fluids may be administered intraperitoneally (or intracoelomically) or intraosseously.

Many exotic pets are anaesthetised for surgery or treatment of painful conditions. The judicious use of analgesics will speed recovery from anaesthesia and illness. Multimodal analgesia is used as the synergistic increase in analgesic potency allows lower doses of drugs to be used, with concomitant lowering of side effects. For example, opioid analgesics are often administered with non-steroidal anti-inflammatory drugs (NSAIDs). Opioids are of particular use when anaesthetising animals, as most also have sedative or tranquillising effects, which will be anaesthetic-sparing.

RECOVERY

If possible, anaesthetic agents should be *reversed*. This will reduce the risk of hypothermia, and also risks associated with cardio-respiratory depression (Erhardt et al., 2000; Henke et al., 1995; Henke et al., 1998; Henke et al., 1999; Henke et al., 2000; Roberts et al., 1993). If part of the anaesthetic protocol that is reversed provided analgesia, for example where opioids are used, consideration should be given to alternative analgesics in the recovery phase.

The postoperative recovery period is often neglected when animals are anaesthetised. In exotic pets, this period is just as important as the anaesthetic time. Patients are still susceptible to many of the risks associated with anaesthesia and a large number of mortalities occur during this time. As many exotic pets are prey species, the recovery *environment* should be quiet and away from predator species that may stress the recovering patient.

The environmental temperature will vary depending on species requirements, but supplemental *heating* is usually necessary until homeostatic mechanisms return. This is particularly important in neonates. Incubators are ideal for this period and also allow the provision of oxygen (Flecknell, 1996). Thermometers are useful to monitor both environmental and patient temperatures, ensuring maintenance of an appropriate temperature.

As with the pre-anaesthetic period, hospital facilities should provide a secure area for patients. Until the animal has recovered enough, soft *bedding*, such as towels or Vetbed® (Profleece, Derbyshire, UK), should be provided, which will not irritate eyes or airways. Water receptacles should be removed until the patient has recovered, to prevent accidental drowning.

Supplemental *fluids and nutrition* are often necessary for a period of time after anaesthesia in exotic pets. This may be directly related to the procedure performed under anaesthesia, but often reflects a state of debility on presentation. Appetite, water intake, urination and defecation should be recorded if possible in the days following anaesthesia. As it is difficult to assess whether many patients have eaten, body weight is recorded daily with all patients (Fig. 1.9).

Depending on the procedure performed or the patient's condition, *analgesia* may be necessary in the period after anaesthesia. Pain and analgesia are poorly understood in many exotic pets, but research suggests that they feel pain and ethics advise that we treat this pain. As with other domestic species, pre-emptive analgesia is preferable. It is often difficult to assess exotic pets for clinical signs associated with pain and clinicians are advised to err on the side of caution, administering analgesics if pain or discomfort may be present. Many species will not show signs of pain as more domesticated species do and signs shown are likely to be subtle. Few exotic pets will vocalise. Animals may be less active than normal, have a reduced appetite and thirst, have an altered appearance, show behavioural changes, or have cardio-respiratory changes (Flecknell, 1996).

Classes of analgesics available for animals include local anaesthetics, NSAIDs and opioids. Most routes, including orally, subcutaneously, intramuscularly, intravenously and epidurally, may be used to provide analgesia. An example of an opioid used in many species is butorphanol, a mixed opioid agonist-antagonist, with primary agonistic activity at the λ-opiate receptor (Vivian et al., 1999). Analgesic effects will vary between species, depending on the presence of the receptor. Meloxicam is a cyclo-oxygenase-2 (COX-2) selective NSAID (Kay-Mugford et al., 2000), available as an injectable formulation or an oral suspension that is easily administered to many animals.

Analgesic drug pharmacokinetics have not been fully evaluated in most exotic pet species and doses often have

not been tested for efficacy. Where analgesic agents have been used in exotic pets to provide pain relief and/or aid anaesthesia, they are discussed in later chapters.

If the patient does not recover in the expected period of *time* for the anaesthetic used and procedure performed, the clinical examination should be repeated. Investigations carried out so far should be reviewed, to identify some aspect of ill health that has been missed. Pending a diagnosis, supportive care should continue with oxygenation, fluids and supplemental heat as required. (The respiratory drive in reptiles is reduced in high concentrations of oxygen, so oxygen supplementation should be provided intermittently in these species.) Monitoring should also be performed continuously until the patient is deemed stable, and then periodically until the animal is sufficiently recovered to be left unattended. The head and neck should be extended to reduce airway obstruction. Laterally recumbent animals should be turned from time to time to reduce passive congestion in the lungs, with the development of hypostatic pneumonia (Flecknell, 1996).

BOX 1.2 Care during the recovery period

- Supplemental heating
- Supplemental oxygen (some cases)
- Comfortable substrate
- Analgesia
- Fluids and nutrition

ANAESTHESIA MONITORING

Guedel described five stages of anaesthesia (Guedel, 1936); more recent reviews consider four stages (Smith and Swindle, 1994). Induction is comprised of stage one (voluntary excitement) and stage two (involuntary excitement). Stage three is surgical anaesthesia, and various reflexes are usually lost at this stage, for example skeletal muscle tone. Stage four is characterised by medullary paralysis, shortly before death. These stages or 'depth' of anaesthesia are assessed using various techniques, mainly physiological parameters and assessment of reflexes. More recent advances have included attempts to monitor 'awareness' during anaesthesia, particularly in human patients (Drummond, 2000).

The depth of anaesthesia is monitored to ensure that the patient is at a sufficient plane for the procedure being performed, and that a fatal overdose does not occur. Other common causes of anaesthetic mortality are equipment problems, hypothermia and cardiovascular collapse (Jones, 2001). Monitoring both patient and equipment throughout anaesthesia and into the recovery period should identify problems early enough to allow appropriate action to avoid fatalities.

Patient monitoring

The stages of anaesthesia described can be difficult to apply across a broad range of species, as responses will vary between animals. Different drugs will also produce anaesthesia in different ways, particularly with regard to *reflexes* or onset time of anaesthesia. Gaseous or intravenous agents produce much more rapid onset compared to intramuscularly administered agents. The depth of anaesthesia required will depend on the procedure to be performed and the patient. Surgical procedures require a deeper plane of anaesthesia than those requiring immobilisation purely for restraint, for example radiography.

The pedal *withdrawal reflex* is a simple way of assessing depth of anaesthesia. The interdigital web of skin is pinched with the limb extended; the tail or ear may be similarly pinched in some animals. At a light plane of anaesthesia, the limb is withdrawn, muscles twitch or the animal vocalises. Eye reflexes and positioning are useful in species such as the pig and primates, where the *palpebral reflex* is usually lost during light surgical anaesthesia with many drugs. However, this reflex is lost at lighter planes with ketamine, and neuroleptanalgesics have unpredictable effects on it. The palpebral reflex is less useful in rodents, and may not be lost until very deep planes of rabbit anaesthesia (Flecknell, 1996).

Most anaesthetics produce *cardio-respiratory* depression. This may include changes in respiratory rate or depth, heart rate and hypotension. Patient monitoring should, therefore, include basic physiological functions, such as respiratory rate and pattern, heart rate and pulse quality. Normal values may not be known for the patient species and anaesthetic combination, but the recording of the above values allows rapid identification of trends that may denote an alteration in the patient's well-being (Flecknell, 1996).

Respiratory system observations will include respiratory rate, pattern and depth. The patient's chest wall may be observed, as may the reservoir bag if the animal is intubated or a tightly fitting facemask is used. A bell or oesophageal stethoscope can be used to auscultate lung sounds. Respiratory monitors may be used to monitor respiratory rate. Some monitors can be used with animals as small as 300 g. A Wright's respirometer can be used to measure tidal and minute volumes, with paediatric versions suitable for animals over 1 kg. Ensure the particular piece of equipment used does not add to dead space or circuit resistance (Flecknell, 1996).

Peripheral *pulses* are extremely useful in monitoring the cardiovascular system, providing an estimation of systemic arterial pressure. These are more easily evaluated in larger mammals, such as rabbits, but difficult in smaller mammals and thick-skinned reptiles. The *capillary refill time* of mucous membranes will be rapid with adequate tissue perfusion. Bell or oesophageal *stethoscopes* can be used to monitor heart rate in most species. *Doppler* blood flow monitors are useful in very small patients and reptiles, as they are able to detect pulses in relatively small arteries (see Fig. 3.8). A decrease in heart rate is usually

associated with a deepening of anaesthesia. Elevations in heart rate often suggest the depth of anaesthesia has lightened, or could be due to pain caused by surgery at an inadequate depth of anaesthesia (Flecknell, 1996).

Techniques for recording ECGs have been reported in several species (Schoemaker and Zandvliet, 2005). The basic principles are the same as for other species, but some allowances are made for difficulties with contact through thick fur or scales. To increase contact, needle electrodes can be used or alligator clips can be attached to subcutaneous needles (see Fig. 12.11). ECG gel is used to enhance electrical conduction. By standardising positioning, ECGs can be interpreted as in other animals. The red (white in the US) cable attaches to the right front leg, the yellow (black in the US) to the left front leg, the green (red in the US) to the left hind leg and the black (green in the US) earth cable to the right hind leg. ECG measurements are reported in various exotic species, some conscious and some anaesthetised (Anderson et al., 1999; Girling and Hynes, 2002; Martinez-Silvestre et al., 2003; Reusch and Boswood, 2003; Whitaker and Wright, 2001). Care should be taken in ECG interpretation as different anaesthetics will affect the results differently.

Assessment of *mucous membrane colour* is a rough measure of blood oxygenation; *pulse oximetry* is a more sensitive technique. Pulse oximeters measure the oxygen saturation in arterial blood; the machines also measure pulse and calculate heart rate. Haemoglobins vary between species, but most human pulse oximeters can be used in mammal species (Allen, 1992; Decker et al., 1989; Erhardt et al., 1990; Vegfors et al., 1991). The probes may be attached to the ear, tongue, foot or tail of patients. Normal oxygen saturation is 95–98% in animals breathing room air, but will increase to 100% when breathing oxygen. Low oxygen saturation correlates with hypoxia and could be due to respiratory depression, airway obstruction, poor contact between the animal and the pulse oximeter, or failure of anaesthetic equipment. If the blood flow falls sufficiently, for example during shock, a signal will not be detected. Small patient size may also reduce the accuracy of values produced, and in these cases trends are more important than absolute values. Machines may also have a high heart rate alarm below the normal rate for a particular species (Flecknell, 1996).

A *capnograph* can be used to measure expired carbon dioxide levels. These machines either sample directly from the anaesthetic circuit (mainstream system) or from a tube close to the endotracheal tube (side-stream system) (O'Flaherty, 1994). The former are more sensitive and give rapid results, but increase dead space in the circuit. For animals with small minute volumes, the expired gas sample may be contaminated with gas from the circuit, giving an underestimation of the end-tidal carbon dioxide; trends are still useful. The maximum value reflects alveolar gas carbon dioxide concentration. The normal range in spontaneously breathing animals is 4–8%. If respiratory failure or rebreathing of exhaled gas occurs, the concentration will increase. Capnographs appear to be less accurate at higher ranges of $P_{ET}CO_2$ (Edling et al., 2001; Teixeria Neto et al., 2002).

Blood gas analysis is the most accurate method of assessing the partial pressures of oxygen and carbon dioxide, blood pH, blood bicarbonate concentration and the base excess. Some analysers can make measurements from 0.1 ml. Changes in body temperature will affect results, and the machine requires calibration for this variable. The main difficulty with this technique is arterial blood sampling. Blood gases are similar for most species. A blood gas carbon dioxide measurement at the start of a procedure can be used to calibrate capnography results (Flecknell, 1996).

ECGs are useful for monitoring the electrical activity within the patient's heart (see Figs 4.9 and 9.8). Electrical activity may continue after the heart stops beating, so ECG output does not always correlate with cardiac output. Machines with an electronic display usually display heart rate also (Flecknell, 1996). Problems may be encountered with the use of ECGs in small patients, where electrode contact may be difficult to maintain.

Assessment of *blood pressure* is an excellent indicator of cardiovascular function. Indirect measurement of systemic arterial blood pressure is possible in many species using a sphygmomanometer, inflatable cuff and Doppler probe, for example using the carpal artery in rabbits (see Fig. 3.8), the ulnar artery in birds (see Fig. 10.3), and the caudal artery in rats. Disadvantages with this non-invasive monitoring are the production of intermittent values, and a failure to detect weak signals when pressure falls. Direct measurements produce a continuous recording, but require arterial cannulation that may not be possible in all species. The femoral artery may be used in rabbits, pigs and larger primates, and the central auricular artery in rabbits. Central venous pressure can be measured via a catheter threaded into a jugular vein and advanced to the anterior vena cava (Flecknell, 1996).

It is important to monitor the *body temperature* of exotic pets during anaesthesia. Temperature homeostasis is reduced during anaesthesia, and inadequate supplemental temperatures rapidly allow body temperatures to fall. Most exotic pets are small animals that succumb readily to hypothermia due to their high surface area to body weight ratio, or are ectotherms and rely on environmental temperature to maintain their metabolic functions. Hypothermia will adversely affect the patient's metabolism, hence prolonging recovery time, and increase the potency of gaseous anaesthetic agents (Regan and Eger, 1967).

Rectal temperature is usually assessed in mammalian species, and is easily monitored using a thermometer (see Fig. 4.7). Care should be taken in species with thin-walled gastrointestinal tracts, such as birds, where cloacal damage may readily occur. Probes for oesophageal placement, skin surface temperature probes, or thermometers for measuring temperature at the tympanic membrane are alternatives. These may not be accurate in all species and should be validated using a conventional thermometer. It is assumed that a reptile's body temperature will equilibrate with the environmental temperature, and for these species an environmental thermometer alongside the patient suffices (Flecknell, 1996).

BOX 1.3 Anaesthetic monitoring

- Reflexes (variations between species and anaesthetics)
- Respiratory system:
  - Rate, rhythm, depth
  - $ETCO_2$ (capnograph)
  - SpO_2 (pulse oximeter)
  - Blood gas analysis
- Cardiovascular system:
  - Heart rate, rhythm (palpation, bell or oesophageal stethoscope, Doppler flow monitor)
  - Peripheral pulses (palpation)
  - Mucous membrane colour
  - Capillary refill time
  - Electrocardiogram
  - Blood pressure (usually indirect method)
- Body temperature

Anaesthetic equipment monitoring

All equipment for the procedure and anaesthetic should be assembled and checked before the animal is induced. The anaesthetic equipment should also be monitored continuously throughout the procedure. Care should be taken to ensure the patient remains connected to the anaesthetic machine, especially if the patient is moved during the procedure. A change in position may also cause the circuit or endotracheal tube to kink, obstructing the patient's respiration. The anaesthetist should be aware of the position of valves in the circuit, ensuring that they are never causing obstruction or excess resistance to the patient's breathing. The pressure regulator dial(s) should be observed to ensure the oxygen supply does not run out, and a spare cylinder should be ready to attach to the circuit if required. Some anaesthetic machines will have an alarm to indicate low oxygen. The vaporiser should similarly be monitored to ensure sufficient volatile agent is present.

Other equipment requiring continuous function assessment includes the patient-monitoring equipment and peripheral devices, such as infusion pumps, if fluids or other drugs are being administered.

RELEVANT TECHNIQUES

Ventilation

Most anaesthetics cause respiratory depression, which may result in hypoxia, hypercapnia and acidosis (Flecknell, 1996). Microatelectasis may occur in the lungs due to reduced tidal volume and perfusion during anaesthesia. It is, therefore, good practice to assist ventilation during anaesthesia. This is facilitated by endotracheal intubation and, therefore, may be limited in smaller patients that cannot be intubated.

Intermittent 'sighing' or PPV of anaesthetised animals helps prevent microatelectasis in the lungs, by inflating the lungs to their normal capacity. PPV also allows the clinician to control oxygen provision to the patient's airways and concentrations of inspired anaesthetic agents. Increasing or decreasing the rate and/or pressure of PPV is one method of lightening or deepening depth of anaesthesia. PPV can either be performed by the assistant, using hand-control of the anaesthetic bag and valve, or mechanically.

Most anaesthetic circuits allow intermittent positive pressure ventilation to be performed by the anaesthetist, but the use of a mechanical ventilator will free the anaesthetist to perform other procedures including patient monitoring. At lighter planes of anaesthesia, spontaneous respiratory movements may interfere with ventilation; neuromuscular blockers will block these movements, but are rarely used in exotic pets (Flecknell, 1996).

Mechanical ventilators apply intermittent positive pressure to the airway and thereby produce controlled ventilation. Mechanical ventilators may be programmed to provide a set number of breaths per minute; they are *'time-cycled'* to switch from inspiration to expiration. *Pressure-limited* machines will deliver gases to a maximum pressure, which is adjustable. This takes account of variability in patient lung compliance, which will change resistance to gas flow. *Volume-limited* machines are adjusted to provide a set volume of gas with each inspiration, and this tidal volume will not be affected by pressure variations. The switch back to inspiration is similarly dependent either on a fixed time interval or a set drop in airway pressure (Flecknell, 1996).

If ventilation is pressure-limited, hypoventilation may occur if the airway becomes occluded or if respiratory compliance reduces. Using a volume-limited machine, an occlusion will cause an increase in pressure that triggers an alarm to warn the operator. Hypoventilation may occur with volume limitation if the anaesthetic system leaks (Edling, 2006).

Providing that an appropriate pressure is selected, the pressure-limited machines are most useful in small exotic pets, as an excessive increase in pressure may lead to damage or even rupture of part of the respiratory tract. A useful safety feature is a pressure relief valve in the circuit between the fresh gas inflow and ventilator, to prevent over-inflation of the respiratory tract (Flecknell, 1996). Examples of mechanical ventilators which may be used in small patients include the SAV03® Small Animal Ventilator ([Fig. 1.10] Vetronic Services, Devon, UK), which can be used in animals from 10 g to 10 kg, or the Nuffield 200® (Penlon Ltd, Abingdon, UK).

The use of a mechanical ventilator allows the anaesthetist to control ventilation reliably, automatically providing intermittent positive pressure ventilation (IPPV). It is extremely useful to be able to set the maximum airway pressure, particularly in small animals where it is easy to over-inflate airways when manual IPPV is performed. Suggested values are presented in later chapters, but are a



Figure 1.10 • Mechanical ventilator for use in animals weighing up to 10 kg (SAV03® Small Animal Ventilator, Vetronic Services, Devon, UK).

guide only, as individual animals may require different pressures to compensate for disease (for example an increased airway resistance due to respiratory pathology). The respiratory rate can also be adjusted appropriate to the species, usually slightly less than the conscious respiratory rate.

As pressures required will vary greatly between species, these are often adjusted in individual cases until the chest (or limbs in chelonia) excursions approximate those normally seen in the conscious animal. Suggested pressures are listed in species chapters; these will vary depending on the weight of the animal, degree of obesity and functional resistance in the airway circuit. Higher pressures are required in large or obese individuals.

Mechanical ventilators can only be used with intubated patients; if used with a loose-fitting mask, the pressure cut-off will never be reached and gas will continuously be infused. The pressure and frequency settings necessary will depend on the species and individual animal. Some species such as rabbits have a very small tidal volume and rapid respiratory rate, while others such as reptiles have a large volume and slow rate. Reptile and avian airways are particularly delicate, and easily ruptured. Animals with airway disease may have an increased lung resistance that necessitates higher ventilator pressures. Observation of thoracic wall movements should allow the clinician to mimic normal inspiratory volumes. End-tidal carbon dioxide levels should be monitored during artificial ventilation, and tightly maintained between 4% and 5% (Flecknell, 1996).

The main disadvantage of using a mechanical ventilator is the requirement for the patient to be intubated, to allow the ventilator to inflate the airways to a specified pressure. Similarly, if the endotracheal tube is too small or there is a leak in the anaesthetic circuit, gas will leak from the system and a normal inspiratory–expiratory pattern will not be producible.

Respiration during ventilation differs from spontaneous ventilation. During spontaneous ventilation, gases are usually inspired during negative pressure in the thorax. When using a ventilator, positive pressure during inspiration will compress the heart and large veins; this may reduce cardiac performance and reduce blood pressure. To reduce this problem, the period of positive pressure should be minimised by increasing gas flow rates, but this should not be allowed to compromise airways using high pressures.

Routes of administration

These are described in more detail in species chapters. The main routes of administration for medications are the same in all species: oral, subcutaneous, intramuscular, intravenous, intraperitoneal (or intracoelomic in avian and reptile species) and intraosseous.

Intramuscular injections are administered in the quadriceps muscles of most animals, although the forelimbs or paravertebral musculature are more commonly used in reptiles. Intramuscular injections in small animals, particularly rodents, may cause muscle damage and pain, and so should be avoided if possible (Wixson and Smiler, 1997). Avoid injections into the caudal thigh, as the sciatic nerve may be damaged (Hedenqvist and Hellebrekers, 2003).

Intraperitoneal access is most commonly used for administration of fluids. Absorption is rapid, but fluids must be warmed to body temperature beforehand to avoid causing hypothermia. Anaesthetic doses necessary are higher when administered intraperitoneally compared to intramuscular or subcutaneous. Doses required to produce the same effect for the latter two routes are 50–75% of that for the former route (Hedenqvist and Hellebrekers, 2003). Drugs administered via the intraperitoneal route are subject to hepatic first-pass metabolism.

Intravenous access can be technically difficult in exotic pets, and in many species sedation or anaesthesia is required. In mammals, the cephalic, saphenous, jugular, auricular and coccygeal veins can be used.

Intraosseous injections are ideal for administration of fluids and emergency drugs, and are used when venous access is not possible (Garvey, 1989). The site for catheter placement varies between species. Aseptic technique is vital for intraosseous catheter placement, with the skin clip and preparation as for surgery. A small needle can be used for an intraosseous catheter, using a piece of sterile surgical wire as a stylet in larger species. The proximal femur or tibia is a commonly used site for intraosseous catheters; the ulna is often used in birds. The limb is grasped in the non-dominant hand, palpating the direction of the bone and the proximal end. The needle is then inserted into the proximal end of the bone. Gentle turning

of the needle with constant pressure will allow the needle to enter through the cortex, with no resistance felt once the medullary cavity is reached.

Confirmation of placement is by injection of a small amount of sterile saline, which should not encounter resistance. Movement of the hub of the needle should move the impregnated bone, and the needle tip should not be palpable in the muscle around the bone. Radiographs can also be used to check the needle site. If intraosseous access is required for a period of time, sterile tubing may be attached for continuous infusion or a heparinised bung may be used as a port. The hub of the needle can be secured using tape and sutures (see Fig. 4.3).

PAIN AND ANALGESICS

Peripheral nerves detecting a noxious stimulus transmit information to the spinal cord and, thence, to the brain. The onset of pain causes physiological changes in the nerves and pain transmission system, leading to increased sensitisation to further noxious stimuli. Inflammation will cause an increased response to a normally painful stimulus; this is peripheral sensitisation. Central sensitisation may also occur, producing a greater and more prolonged response to stimuli (Paul-Murphy, 2006; Woolf and Chong, 1993). This sensitisation may persist for some time after the initial noxious stimulus is removed.

Provision of analgesia is usually twofold. Pre-emptive analgesia administered before pain occurs will reduce the 'windup' described above as peripheral and central sensitisation (Woolf, 1994; Woolf and Chong, 1993). Secondly, the use of different classes of analgesics or multi-modal analgesia will affect the pain transmission and perception at several points in the physiological pain pathway. A synergistic effect may be seen when two or more analgesics that act via different mechanisms are used in combination. This is important when ill animals may succumb to side effects more easily, and enables lower doses of individual drugs to be used to produce the same analgesic effect. Tranquillisers in some anaesthetic protocols may reduce anxiety and potentiate the analgesic effect.

Analgesics

Preoperative administration of *local anaesthetic* agents, such as lidocaine (lignocaine) and bupivacaine, will prevent or attenuate 'windup'. These agents are often prepared with adrenaline (epinephrine) to reduce absorption systemically. Local anaesthetics are most commonly administered via a splash block, a local line block or regional infiltration. A line block involves subcutaneous injection into tissue along the site of an incision.

Opioid receptors occur in the central and peripheral nervous systems. The three classes of receptor involved in analgesia are mu (μ), delta (δ), and kappa (κ). Species variation exists in the locality, number and function of these receptors. Opioid drugs have morphine-like effects, likely mediated via an increase in serotonin synthesis (Paul-Murphy, 2006). Most opioids produce sedation and respiratory depression as well as analgesia. Different drugs act on different receptor classes and will produce different effects.

Eicosanoids such as prostaglandins and thromboxane are released when tissues are injured, resulting in inflammation and nerve ending sensitisation (Paul-Murphy, 2006). *NSAIDs* inhibit COX enzymes, thus interfering with eicosanoid synthesis. By reducing these products, NSAIDs decrease inflammation and modulate CNS effects. The expression of COX-1 and COX-2 enzymes varies between species. NSAIDs can be utilised to treat many types of pain, including musculoskeletal and visceral, as well as acute or chronic pain.

Side effects may be seen with NSAIDs, as they may affect renal, hepatic or gastrointestinal systems. These drugs are, therefore, used with caution in animals with pre-existing disorders of these systems or in hypovolaemic animals where renal blood flow may be reduced. It is not known if similar side effects will be seen in all animals, but renal lesions have been reported with NSAID use in both mammals and birds (Ambrus and Sridhar, 1997; Klein et al., 1994; Lulich et al., 1996; Orth and Ritz, 1998; Radford et al., 1996). Little research has been done on the use of these drugs in exotic pets. Drug doses are often extrapolated and therapeutic serum levels are not known for most species.

SPECIAL CONDITIONS

The choice of anaesthetic for *pregnant* animals should consider the consequences of the drugs on the fetus(es) as well as the dam. In general, gaseous agents, such as isoflurane, are used where possible, as recovery does not rely on drug metabolism. Positioning should ensure uterine contents do not put excess pressure on the thoracic region, which may impede respiration. Oxygen, heat and fluids should be supplemented; this will avoid hypoxia, hypothermia and hypotension respectively. The dam should not be fasted, blood glucose should be monitored and hypoglycaemia treated. If injectable agents have been administered as part of the anaesthetic, give reversal agents to neonates delivered via Caesarean as well as to the dam. Doxapram is also useful in stimulating respiration in these neonates (Flecknell, 1996).

Neonates are more susceptible to many of the problems associated with anaesthesia, such as hypothermia and hypoglycaemia. Cardio-respiratory function and drug metabolism are also likely to be reduced compared to adult animals. Inhalational agents are frequently used if neonatal anaesthesia is to be performed. Higher concentrations of agents are often required to anaesthetise neonates (Flecknell, 1996).

EMERGENCY PROCEDURES AND DRUGS

In most instances, monitoring procedures will detect early signs of problems during anaesthesia. If the patient is stable, there will be minimal changes in parameters being measured. However, there may be times when more aggressive responses and intervention are required.

Respiratory problems

Anaesthesia normally results in respiratory depression, but a significant reduction in respiratory rate is likely to be associated with problems. Onset of respiratory failure may be indicated by a reduction in respiratory rate, for example in rabbits and rodents to less than 40% of the unanaesthetised rate, or a fall in tidal volume (Flecknell, 1996).

If ventilation is not assisted, the respiratory depression during anaesthesia will result in an increase in partial pressures of carbon dioxide. Dead space will allow rebreathing of expired carbon dioxide and further increased levels. If this persists for prolonged periods, hypercapnia and acidosis will result. IPPV, or 'sighing' the patient periodically, will reduce this build up of carbon dioxide (Flecknell, 1996).

An increase in respiratory rate is likely to correspond to a lightening of anaesthesia, but may also occur in hypercarbia. Hypercarbia will result in a gradual rise in end-tidal carbon dioxide concentration, as exhaled gas is rebreathed. This may be due to a lack of fresh gas, soda lime exhaustion or anaesthetic circuit problems. A decrease in end-tidal carbon dioxide may be due to increased ventilation, hypotension or reduced cardiac output. The carbon dioxide waveform can be interpreted further, with sudden reductions indicating airway obstruction, disconnection of breathing circuit from the animals or cardiac arrest (Flecknell, 1996).

Hypoxia will result in cyanosis of mucous membranes, but only with very low oxygen saturations (less than 50% in most species). Pulse oximetry is a more accurate technique for monitoring blood oxygen saturations, with a drop of 5% or more requiring action. Hypoxia below 50% is life-threatening (Flecknell, 1996).

Inadequate gas exchange results in a decrease in blood oxygen and/or an increase in carbon dioxide concentration (Flecknell, 1996). Blood gas analysis is not routinely performed in small patients, and the reader is referred to other texts for more detailed blood gas analysis interpretation (Martin, 1992).

If respiratory failure is identified, the patient and equipment should be checked. Ensure that oxygen is being supplied (i.e. that oxygen remains in the cylinder or circuit, and that the circuit is still attached to the patient and is unimpeded). Switch off volatile anaesthetic agents and/or administer reversal drugs for injectable agents. One hundred per cent oxygen should be administered, performing positive pressure ventilation if possible. In small unintubated animals, breaths can be forced by thoracic compression. (In reptiles, administration of 100% oxygen will depress ventilation.)

Where bronchial secretions build up during anaesthesia, they may obstruct small airways. *Anticholinergics*, such as atropine and glycopyrrolate, can be used to reduce secretions. Humidification of inspired gases using nebulisers can reduce drying of the secretions, allowing them to flow more freely and reducing the risk of obstruction. This is less important for short procedures, but more so for longer anaesthetics or for dyspnoeic animals in oxygen chambers (Fig. 1.11).



Figure 1.11 • Humidifiers can be used to reduce drying of respiratory passages by gases in animals requiring supplemental oxygen.

Doxapram is a respiratory stimulant, available in both injectable and topical forms (Dopram-V®, Willow Francis). It may be used to treat anaesthetic-associated respiratory arrest or to counteract the respiratory depressive effects of fentanyl. Doxapram may also reverse fentanyl's analgesic properties (Flecknell et al., 1989). Doxapram's duration of activity is 15 min (Cooper, 1989) and repeated administration may be necessary.

Cardiovascular problems

These often result from anaesthetic overdose, but may also be secondary to respiratory failure causing hypoxia and hypercapnia, following severe blood loss, or hypothermia. Circulatory failure may result in delayed capillary refill time, with blanched mucous membranes if associated with hypovolaemia, low peripheral temperature compared to rectal temperature, hypotension and variable (increased or decreased) heart rate (Flecknell, 1996).

If possible, the patient should be intubated to allow positive pressure ventilation with 100% oxygen. If intubation is not possible, a facemask should be used to provide oxygen while chest compressions are used to ventilate the lungs. External cardiac compressions should also be performed if cardiac arrest has occurred. Pre-placement of an intravenous catheter at induction provides venous access in such an emergency, allowing administration of reversal agents or other drugs if necessary. Atropine has parasympatholytic

effects; by stimulating supraventricular pacemakers, it may correct supraventricular bradycardias or a slow ventricular rhythm (Edling, 2006). Adrenaline (epinephrine) is a positive inotrope; it initiates heart contractility, increases heart rate and cardiac output. Atropine should be administered if complete heart block or bradycardia is present, lidocaine if fibrillation or arrhythmia has occurred, and adrenaline (epinephrine) if asystole is present. Fluid therapy is important if hypovolaemia is present (Flecknell, 1996).

Other problems

Hypothermia is unlikely to be an acute problem and should be prevented by close monitoring of body temperature and provision of supplemental heating. If it occurs the patient should be slowly warmed using heat sources as described above. Warmed fluids should be administered. The recovery time will be prolonged and ventilatory support is likely to be required for a longer period.

Vomiting and regurgitation are possible in some species. (Significantly, they are not possible in rabbits and rodents.) If they occur, immediate action should be taken to reduce the risk of inhalation of gastric contents that may cause an immediately fatal respiratory obstruction or lead to aspiration pneumonia. The presence of an endotracheal tube will help protect the airways from these problems. The animal's head should be lowered and material swabbed or aspirated from the oral and pharyngeal cavities (Flecknell, 1996).

BOX 1.4 Emergency procedures

- Intravenous access (better to have it before you need it!):
  - Fluids for shock, hypovolaemia
  - Blood transfusion for severe blood loss
- Airway/breathing:
  - Oxygen via endotracheal or transtracheal intubation, nasal catheter, or facemask
  - Ambu-bag or resuscitator for PPV (Fig. 1.12)
- External cardiac massage
- Drug administration:
  - Adrenaline (epinephrine)
  - Atropine, glycopyrrolate
  - Doxapram
  - Diazepam

If an unintubated anaesthetised animal suffers from apnoea, two methods can be used to induce inspiration and expiration artificially. IPPV can be instigated with a tight-fitting facemask. There is a possibility of inflating the oesophagus and stomach using this technique, causing iatrogenic bloat. The other technique is most effective in mammals (which possess a diaphragm), and involves rocking the patient along the body length so that the abdominal viscera move towards (inducing expiration) and away from (inducing inspiration) the lungs. This is obviously more difficult in a patient undergoing surgery, for example a coeliotomy, where large body movements may not be possible.



Figure 1.12 • Resuscitators are available for use with small patients.

BOX 1.5 What to keep in your crash box

- Adrenaline (epinephrine)
- Atropine
- Doxapram (drops and injectable)
- Diazepam
- Endotracheal tubes (uncuffed), 1–6 mm diameter
- Glycopyrrolate
- Intravenous catheters (20–26 gauge)
- Laryngoscope, with size 0–1 Wisconsin blade
- Local anaesthetic spray
- Local anaesthetic ointment (for example lidocaine with prilocaine, EMLA®)
- Needles (18–24 gauge) and syringes (1–5 ml)
- Ocular lubricant
- Penlight
- Adhesive tape

CHAPTER OUTLINES

The remainder of the text is divided according to taxonomic groups, with chapters on mammals, reptiles, birds, amphibians, fish and invertebrates. An introductory section will describe group anatomy and physiology that is relevant to anaesthesia, along with an overview of techniques appropriate for those species. Although some basic husbandry information and veterinary medicine is provided where

Table 1.2: Doses of emergency drugs (doses vary between species and may require to be repeated) (see Fig. 1.8)

DRUG	DOSE (mg/kg)	ROUTE	INDICATION/COMMENT
Adrenaline (epinephrine)	0.02–0.20	IM, IV, IT, SC	Cardiac arrest (fibrillating or asystole) Dilute before use in small patients
Atropine	0.01–0.04 (mammals) 0.2 (birds, reptiles) 0.1 (amphibians, fish)	IM, IV, SC	Cardiac arrest (heart block, bradycardia). Ineffective in animals with atropinesterase (e.g. rabbits)
Dexamethasone	1–2	IM, IV, SC, PO	Ferrets <8 mg/kg, birds <6 mg/kg
Diazepam	0.5–5.0	IM, IV, IP, IO	Seizures
Doxapram	5	IM, IV, IP/ ICe, SC	Short duration of effect, may require repeated dosing (typically every 15 min)
Frusemide	1–10	IV, IM	Diuretic for oedema, pulmonary congestion, ascites
Glycopyrrolate	0.01–0.02	SC, IM, IV	Bradycardia Alternative to atropine for animals with atropinesterase
Lidocaine (lignocaine)	1–2	IV, IT	Cardiac arrest (fibrillating)

Key: ICe = intracoelomic, IM = intramuscular, IO = intraosseous, IP = intraperitoneal, IT = intratracheal, IV = intravenous, SC = subcutaneous

(Carpenter, 2005; Flecknell, 1996)

relevant for anaesthesia and the peri-anaesthetic period, it is not possible to cover these areas in detail; the reader is referred to other texts for further information on these topics. Pathologies are briefly mentioned, to outline common problems that may affect anaesthesia. Within each of the three larger sections (mammals, birds, reptiles), subsections will discuss different families, for example lizards, snakes, chelonia and crocodilia. Each subsection will provide further detail on these families and describe anaesthesia with drug doses and technical procedures specific to those animals, for example intubation techniques.

The aim of the chapters is to make the clinician aware of problems common to each species, to guide pre-anaesthetic preparations. Where pathologies may affect the choice of anaesthetic, protocols are suggested for certain cases.

Many procedures have not been formally reported in exotic pets, but medicine from more common species can often be applied. Where possible, known drug doses are given, but most drugs are not licensed for use in exotic animals. Pet owners should be informed and, ideally, written consent obtained to use drugs off-label.

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2 Mammal anaesthesia

INTRODUCTION

A wide variety of mammals are kept in captivity as pets and presented to the veterinary surgeon for different reasons. This chapter will cover those mammal species of exotic pet commonly presented to veterinary practices. Sedation or anaesthesia may be required for examination (for example, African pygmy hedgehogs – *Atelerix albiventris*), phlebotomy (for example, guinea pigs – *Cavia porcellus* and some ferrets – *Mustela putorius furo*), imaging (ultrasonography, radiography, CT, MRI) or surgical procedures (for example dentistry, wound repair, neoplastectomy or neutering).

Veterinary practitioners are often wary of anaesthetising small mammals due to the risks, real and perceived, of associated morbidity and mortality. A sound knowledge of species-specific anatomy and physiology, and application of basic principles can greatly reduce these risks. However, much individual variation exists in response to anaesthetics in these animals. Patient health status and the procedure to be performed under anaesthesia have been shown to be significant factors in anaesthetic-related deaths (Brodbelt et al., 2005). Veterinary assessment of the patient's condition should be considered before embarking on a 'routine' anaesthetic regime, particularly where injectable agents are used, when it may not be possible readily to alter effects of the anaesthetic should problems arise.

Small mammal species seen in veterinary practice comprise several families, most of which are herbivorous, but others are omnivorous, insectivorous or carnivorous. Species differences will be discussed along with generalisations that will aid anaesthesia across the groups. This chapter will discuss anatomy and physiology pertinent to anaesthesia in small mammals. Later subsections cover the veterinary clinician's approach to individual cases, discussing how to minimise risks associated with anaesthesia. A choice of anaesthetic protocols will be described, to allow clinicians to make an informed choice for their patient.

ANATOMY AND PHYSIOLOGY

Many factors will affect how patients respond to anaesthetics. Some anatomical and physiological factors will affect how anaesthesia is approached and maintained in different animals. General factors are discussed in this section, with species-specific sections in later chapters.

Stress

The primary factor affecting hospitalised small mammals is stress, particularly for prey species, such as rabbits and guinea pigs. Loud noises to which the patient is not accustomed and the presence of predator species in close proximity (within sight, hearing or smell of the prey animal) will cause stress. Prey species should, therefore, be hospitalised in a separate kennel area to predators (ferrets will come into this latter category), where they cannot see, hear or smell predator species. The environment should be quiet, with subdued lighting for nervous individuals, and the temperature maintained appropriately warm (Table 2.1).

Stress will cause adrenergic stimulation. Changes may occur in the animal's cardiovascular (hypertension), renal (reduced renal perfusion) and gastrointestinal systems. These may impact on the patient's response to anaesthesia.

Respiratory system

Respiratory tract anatomy differs somewhat in these small mammals. In rodents and lagomorphs, the larynx is situated dorsally within the oropharynx, closely associated with the nasopharynx (Fig. 2.1), making the animals obligate nasal breathers (Vaughan, 1986). This and the small diameter of the upper airway mean that intubation is readily possible in only a few small mammal species, including the rabbit, ferret and non-human primates.

Table 2.1: Physiological information for some common species (conscious values)

SPECIES	ADULT BODYWEIGHT	RECTAL TEMP (°C)	USUAL ENVIRONMENTAL TEMPERATURE (°C)	HEART RATE (BPM)	RESPIRATORY RATE (BPM)
African pygmy hedgehog[7]	250–600 g (males double female size)	36.0–37.4	23–32 (optimum 24–29)	180–280	25–50
Chinchilla[6]	400–600 g (female larger)	37–38	18.3–26.7 (optimum 10–20)	100–150	–
Chipmunk[8]	72–120 g	38 (or a few degrees above environmental temperature when hibernating)	–	–	75
Common marmoset[12]	350–400 g	39–40	–	200–350	50–70
Ferret[3]	Average 600 g (female) –1200 g (male)	37.8–40	–	200–400	33–36
Gerbil[10]	70–120 g	37.0–38.5	–	300–400	90–140
Guinea pig[2]	750–1200 g (male larger)	37.2–39.5	18–26	190–300	90–150
Mouse[10]	25–63 g (female larger)	37.5	24–25	500–600	100–250
Pig[1]	40–200 kg (breed-dependent)	38.4–40	10–32	70–80	20–30
Prairie dog[4]	0.5–2.2 kg (male larger)	35.3–39.0	20–22	83–318	–
Rabbit[9]	1.0–10 kg (depending on breed)	38.5–40.0	15–21	180–300	30–60
Rats[10]	225–500 g (male larger)	38	18–26	260–450	70–150
Sugar glider[11]	80–160 g (males larger)	32 (cloacal temperature); 36.3 (rectal temperature)	–	200–300	16–40
Syrian hamster[5]	85–150 g (female larger)	–	20–24	280–412	33–127

[1] (Braun and Casteel, 1993; Straw and Merten, 1992; Taylor, 1995); [2] (Flecknell, 2002); [3] (Fox, 1998; Lewington, 2000; Schoemaker, 2002); [4] (Funk, 2004; Long, 1998; Tell, 1995); [5] (Goodman, 2002); [6] (Hoefer and Crossley, 2002); [7] (Ivey, 2004); [8] (Meredith, 2002); [9](Meredith and Crossley, 2002); [10] (Orr, 2002); [11] (Fleming, 1980; Johnson–Delaney, 2002); [12] (Thornton, 2002)

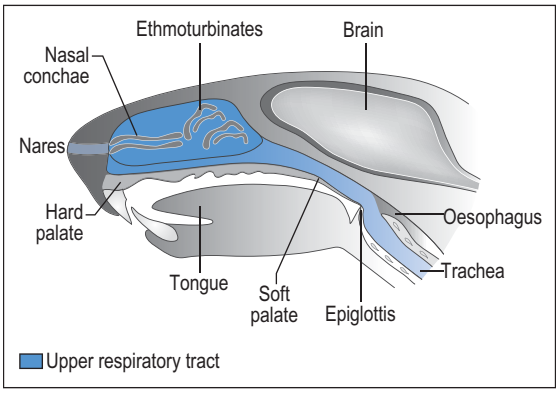


Figure 2.1 • Upper respiratory tract in a typical nasal breather (rat). (After O'Malley, 2005)

These adaptations to increased airflow make small mammals particularly susceptible to respiratory tract disease. Many pet animals are also exposed to husbandry conditions that increase their susceptibility to disease; for example, stress associated with overcrowding or poor nutrition leading to immune compromise, inappropriate temperatures and ventilation, or respiratory irritants, such as ammonia build-up from urine in unclean bedding, or the volatile oil thujone in cedar or pine shavings (Brown and Rosenthal, 1997; Orr, 2002). In some cases, respiratory disease is subclinical. Common causes of pneumonia include *Pasteurella multocida* in rabbits and *Mycoplasma pulmonis* in rats, which result in a reduction in respiratory capacity. While these changes may not cause clinical signs in the conscious patient, the depressant effects of anaesthesia may further compromise the respiratory system and lead to a potentially life-threatening situation. The clinician should, therefore, use the history and clinical examination to try to identify husbandry conditions that may predispose or aggravate respiratory disease, as well as previous problems in the history that may have resulted in consolidation of lung tissue and reduced function, and current clinical disease.

Urinary system

Urine output should be monitored in animals undergoing anaesthesia. Although catheterisation is usually not possible, a rough estimate of urine production can be performed by weighing bedding material. This is particularly useful if renal disease is suspected. Incontinence pads are weighed before use (checking that the patient does not ingest them) and reweighed after use; 1 ml of urine will weigh approximately 1 g.

Digestive system

Similarly, close attention should be paid to appetite and faecal output. A major concern primarily in herbivorous species, such as the rabbit, guinea pig and chinchilla, is that of gastrointestinal hypomotility (ileus) during hospitalisation and post-anaesthesia. Adrenergic stimulation caused by stress will reduce gastrointestinal motility and predispose ileus (Harcourt-Brown, 2002b).

Poor positioning in species such as rabbits during anaesthesia may allow the large gastrointestinal tract to put pressure on the diaphragm, resulting in respiratory dysfunction. Rodents and lagomorphs cannot vomit (due to curvature of their stomach) and so fasting is not required. Ferrets can vomit and so should be fasted for at least 4 h before anaesthesia. Most other small species are not fasted, for example sugar gliders, due to the risk of hypoglycaemia. Larger species such as minipigs are routinely fasted.

Body size

The mammals to be considered here are, in general, smaller than most species being anaesthetised by veterinary surgeons in practice. An exception would be the larger species of rabbits, such as giant breeds that weigh over 5 kg. Small mammals will have a greater surface area to body weight ratio, with an associated high metabolic rate and energy intake (Hurst, 1999). This increases their susceptibility to hypothermia, dehydration, hypoglycaemia and hypoxia (O'Malley, 2005).

There is also a much greater possibility of overdosing with injectable medications in small patients. This risk can be reduced by accurately weighing the patient on electronic scales (see Fig. 1.9), accurate to 0.1 kg for larger species such as rabbits and to 1 g for small rodents, before administration of anaesthetic drugs. Obviously some drug volumes will be minute; in this case, the use of insulin syringes or dilution of drugs before administration will reduce the risk of overdose. If syringes with separable needles are used, the drug volume in the needle hub may be relatively substantial and should be considered when mixing drugs.

Small body size is associated with a higher oxygen demand, for which an increased oxygen intake is required. Rabbits and rodents have comparatively small lungs, but increase airflow through their respiratory tract using their high chest wall compliance and vital capacity, along with low residual lung capacity. Higher oxygen intake is also improved with short airways and high respiratory rates. Oxygen exchange is facilitated by many alveoli with thinner diameter (for example, 35–75 μm in the Syrian hamster compared to 200 μm in the cat) (Donnelly, 1990).

Systemic disease

Certain conditions visible locally on external surfaces may have concurrent systemic disease (for example, lung metastases from uterine adenocarcinomas in rabbits [Greene and Saxton, 1938] or mammary carcinomas or adenocarcinomas in mice). Systemic disease (for example, renal or hepatic impairment, and septicaemia) may be difficult to detect in small animals. Larger species, such as rabbits, may readily be blood-sampled or imaging modalities used to assess before anaesthesia, while small animals, such as hamsters, are likely to require anaesthesia to perform these investigative procedures. For this

reason, history and clinical examination form a much greater part of pre-anaesthetic assessment and decision-making in smaller than in larger species.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

Pre-anaesthetic assessment of the patient is vital, as it may highlight potential problems or identify disease processes that may affect anaesthesia. A complete history of the animal should include husbandry details, which may have altered through the pet's lifetime, and any previous illnesses or clinical signs noted by the owner. The animal should be observed in its carrying container or a kennel for signs of dyspnoea or other illness that the owner may have missed. Small rodents (including rats, mice and gerbils) may have oculo-nasal porphyrin staining in response to stress or illness. Handling many of the mammal species discussed in this chapter will change basic physiological data; for instance, heart and respiratory rates are likely to be elevated. The clinician should be familiar with manual restraint of species, in order to reduce stress during clinical examination and preparation for anaesthesia. Readers are referred to other texts for handling techniques. Animals with cardio-respiratory compromise should be handled with great care and as little as possible, to avoid compromising the patient further.

A full clinical examination should be performed for every patient, and most small mammal pets are amenable to conscious veterinary examination. An exception may be the non-human primate that is not routinely handled, but even in these a pre-anaesthetic examination should be performed to assess cardio-respiratory function. Abdominal palpation may identify problems, such as space-occupying masses, for example neoplasia (or associated pulmonary metastases affecting lung function), which may not be causing clinical signs in the conscious animal, but may reduce respiratory function by reducing diaphragmatic movement when anaesthetised. Historical or clinical findings may identify disease processes and further investigation, such as blood tests or ultrasonography, may be warranted before anaesthesia is induced.

Blood analysis may be required to further evaluate disease processes and metabolic function. In general terms, approximately 10% of the blood volume may be removed in a healthy individual without adverse effects, allowing 3 or 4 weeks to recover before repeated venepuncture. Total blood volumes vary for different species. Obviously this volume may be altered if the animal is ill or already hypovolaemic.

Supportive care and choice of anaesthetic

Findings from investigative techniques should be taken into consideration, and the anaesthetic protocol selected and adjusted as necessary. Sedation or gaseous anaesthesia may be necessary for some investigative procedures, such as radiography, and the benefits to be gained from information should be balanced against the risks of sedation or anaesthesia in the animal. For some patients, anaesthesia should be postponed until the patient can be stabilised with medical treatment of illness or fluid and nutritional support for dehydration and debilitation. Use of anaesthetic agents that may have cardiovascular effects in a dehydrated patient may lead to circulatory failure (Flecknell, 2006).

Unless they are presented for prophylactic procedures (for example, ovariohysterectomy), most pets are unwell and often debilitated. The patient's history and clinical examination should allow the clinician to triage the animal and decide whether it is fit for an anaesthetic. The animal's condition should be stabilised if necessary before anaesthesia, for example by administration of fluids, nutritional support and warmth. Nutritional and fluid supports are discussed in more detail in later sections, but should aim to provide a diet similar to that normally given in a readily digestible form. Many proprietary brands of supplemental nutrition are available. Other medications, such as analgesics or antibiotics, may be required in certain circumstances.

An accurate weight is essential for small patients, particularly if injectable agents are to be used. Most agents can be 'topped-up' if the level of sedation or anaesthesia is insufficient for the required purpose, but many cannot readily be reduced or reversed. Exceptions to this are inhalational agents where the vaporiser setting can be changed and inspired percentage of anaesthetic agent reduced; medetomidine that can be reversed with atipamezole, opioids (for example fentanyl) that can be reversed with partial agonists/antagonists (such as butorphanol and buprenorphine), and diazepam or midazolam with flumazenil.

EQUIPMENT REQUIRED

A trained *assistant* is vital for assisting with anaesthesia induction and monitoring anaesthetic maintenance while the clinician performs the procedures required. It is preferable to have an anaesthetist who can stay with the animal throughout the procedure. This is a good reason for preparing all equipment necessary prior to induction of anaesthesia.

Appropriate sized and shaped *facemasks* should be used, for example small cat masks with pliable soft vinyl (Harvard Apparatus, Holliston, MA), rodent masks with a clear cone for full visualisation and flexible, replaceable rubber diaphragm (VetEquip, Pleasanton, CA), or circuits with flared nose end to create a facemask (VetEquip, Pleasanton, CA). Clear facemasks are excellent for visualisation of mucous membrane coloration during anaesthesia (Harcourt-Brown, 2002a).

The mask should not be too large for the animal, as this will create dead space within the mask. Dead space could be 40 ml or more with facemasks routinely used in species such as rabbits (Bateman et al., 2005). Facemasks should



Figure 2.2 • Fluovac® active scavenging system (Harvard Apparatus, Kent, UK)

Table 2.2: Suggested ventilation rates for mammals

SPECIES	BREATHS PER MINUTE
Guinea pig	50–80
Pig	15–25 (<20 kg), 10–15 (>20 kg)
Primate	40–50 (<5 kg), 10–30 (>5 kg)
Rabbit	25–50
Rat	60–100
Other rodents	80–100

(Adapted from Flecknell, 1996)

also be tightly fitting, to reduce escape of anaesthetic gases into the workplace environment. Active scavenging will reduce environmental contamination, for example the Fluovac® (Harvard Apparatus, International Market Supply, Congleton, UK) supplies anaesthetic gases and simultaneously scavenges (Fig. 2.2).

A selection of uncuffed endotracheal tubes should be maintained for *intubation*, with sizes from 1.5 to 5.0 mm for rabbits, ferrets, and small non-human primates. Intravenous over-the-needle catheters can be used to intubate rodents (60 mm size 14 for guinea pigs, 55 mm size 16 for hamsters, and sizes 14–20 for rats), but the technique is difficult and not routinely performed. It is vital with these small diameters of tubes to ensure they are free of obstructions, and should be cleaned thoroughly and disinfected between patients. Before use, the patency of the tube should be checked, for example by blowing or passing gas from an anaesthetic machine through it. Any build-up of secretions or other material may readily obstruct small tubes and lead to a fatal airway blockage in the anaesthetised patient, or at the very least substantially reduce air flow and pulmonary ventilation leading to hypoxia.

A *laryngoscope*, otoscope or small endoscope is useful for intubation of rabbits, ferrets and non-human primates. A small laryngoscope blade of size 0 or 1 will allow access to most oral cavities. Gags and cheek dilators may also aid visualisation, for example to allow examination or cleaning of the oral cavity after induction.

Since many of these species are obligate nasal breathers, soft *nasogastric catheters* are useful for administration of oxygen where tracheal intubation is not feasible.

As discussed above, many of these species have a small lung capacity and low tidal volume. It is thus imperative to use *anaesthetic circuits* with low dead space. A T-piece (see Fig. 1.1) or mini-Bain (for example, the rodent non-rebreathing circuits with nosecone; VetEquip, Pleasanton, CA) circuit will suffice for most animals.

Mechanical *ventilators* are of great use in intubated animals. Many can be calibrated for use in very small animals (for example the BASi Vetronics® small animal ventilator [see Fig. 1.10] may be used in animals weighing as little as 10 g and as much as 10 kg).

TECHNIQUES

Routes of administration

It is beneficial to consider the small size of many mammal patients when administering medications, particularly via the intramuscular or intravenous routes. Excessively large volumes may lead to muscle necrosis or volume overload, respectively. Anaesthesia with injectable agents often consists of relatively large volumes and should be divided between multiple intramuscular sites.

Ventilation

Mechanical ventilators can only be used with intubated patients. The pressure settings on mechanical ventilators will vary between species. The most valuable guide is visualisation of the patient as gases are forced into the lungs; thoracic wall movement should be similar to that seen in a normal patient. Similarly, respiratory rates should be the same as the animal's normal respiratory rate. This may need to be increased in order to increase anaesthetic depth.

It has been shown that prolonged mechanical ventilation may cause lung parenchymal inflammation. This effect is worse at high-inflation flows (D'Angelo et al., 2004).

ANAESTHESIA MONITORING

The anaesthetist should continuously observe many facets of the anaesthetic. This includes the anaesthetic machine and circuit, the patient, and the clinician. If a painful

procedure is being performed, a deeper plane of anaesthesia will be required than when a non-manipulative procedure is being undertaken.

Observations on the patient

Positioning

This is of great importance during anaesthesia. As already mentioned, airways in many of these animals are narrow and easily occluded. Many of these species are obligate nasal breathers and the nares should be kept clear. The neck should be extended to align the nasal or oral cavity with the trachea. This is necessary even if the animal is intubated, as the small endotracheal tubes used may kink if the neck is flexed. The assistant should also monitor the proximal end of the endotracheal tube to ensure it does not kink and become occluded between the patient and the anaesthetic circuit. The anaesthetic circuit should be attached to the endotracheal tube firmly and monitored for disruption. This may happen, for example, during repositioning for a new procedure after induction. Many herbivorous species have small lungs and large abdominal viscera; the animal should be tilted so the thorax is slightly higher, to reduce pressure on the diaphragm, which may impede respiration. Care should also be taken not to compress the thorax with equipment (Redrobe, 2002).

Cardiovascular system

The cardiovascular system should be monitored. Heart rate and rhythm should be continuously assessed. In larger animals, an oesophageal stethoscope is most useful, but in smaller species a bell stethoscope may be used against the thoracic wall. In some cases, a Doppler flow detector device may be used to auscultate the heart. The femoral artery is palpable in most patients. Peripheral pulses can also be palpated in larger species, such as the rabbit, for example the central auricular and metatarsal arteries (Reusch, 2005).

Assess the colour of the anaesthetised animal's mucous membranes. The most readily accessible membranes are those of the oral cavity or the tongue. If pulse oximetry or capnography is not being used, a change in membrane colour to blue or grey may be the first sign of airway obstruction or other cause of reduced oxygen supply in the patient's circulation.

Respiratory system

Observe the animal's respiratory rate, depth and rhythm. It may be possible to observe movements of the patient's thoracic or abdominal walls, but these may be obscured if the animal is draped for surgery. The use of clear plastic drapes is to be recommended, allowing better observations of the patient. If the animal is intubated, it should be possible to observe movements of the reservoir bag with respiration. These may also be visible if a close-fitting facemask is used. If there are leaks in the anaesthetic system, it is unlikely that the reservoir bag will move with each of the animal's breaths.

Central nervous system

Trends in heart rate, and respiratory rate and depth are useful to assess anaesthetic depth, along with monitoring of reflexes. Species variations will exist, but reflexes are similar to dogs and cats. The toe pinch is more reliable in the hindlimb of most species. Other reflexes that may be used are the palpebral, corneal, level of muscle relaxation including jaw tone and response to surgical stimuli.

Anaesthetic monitoring equipment

Pulse oximetry

This can be useful, but may be unreliable in some animals. For larger species, such as rabbits, the sensor may be sited on the tongue or the ear (Harcourt-Brown, 2002a). The base of the tail may be useful, but if thickly furred requires clipping. The probe can be attached to the feet in small animals, but is not useful for animals with haired feet (including rabbits and Russian hamsters, *Phodopus sungorus*). The pulse oximeter is useful to detect trends in oxygen saturation, but poor contact may reduce accuracy. Anaesthetic agents that reduce the peripheral circulation, for example medetomidine or ketamine, may affect the quality of the signal (Harcourt-Brown, 2002a).

Electrocardiography

Electrocardiogram (ECG) pads can be placed on the patient's feet if hairless; otherwise (for example, in rabbits) use filed-down crocodile clips on skin for short-term recording (Reusch and Boswood, 2003), or clip small area lateral hocks and elbows for application of ECG pads and longer recording. Remember that this will only record electrical activity in the heart and not mechanical function.

Respiratory monitors

These may be used in larger species, but increase the resistance to breathing significantly in smaller animals.

Capnography

This can be used in many species, but care should be taken with small patients that the capnograph does not add to dead space within the circuit or increase circuit resistance.

Thermometers

Core body temperature can be assessed using thermometer probes. Most practices have rectal thermometers, with digital thermometers being more accurate. Some digital thermometers will have a remote sensor, which is of great use when surgical drapes may cover the perineal area. Oesophageal probes may also be used in larger species (Harcourt-Brown, 2002a).

PERI-ANAESTHETIC SUPPORTIVE CARE

Minimise anaesthetic time

Despite accurate anaesthetic dosing and careful monitoring of the anaesthetised patient, any anaesthetic will depress normal metabolic functions. This includes thermoregulation and, often, cardio-respiratory function. The anaesthetic time can be minimised by preparing all drugs and equipment before inducing anaesthesia, in order to reduce the risk to the patient.

Hospitalisation facilities

Supplemental *heat* is usually required for anaesthetised patients and during the recovery period.

Always supplement *oxygen*, even when using injectable anaesthetic agents (many may cause depression of the cardio-respiratory system). This may be via an endotracheal tube, a laryngeal airway mask (see rabbit and pig sections), a facemask, a nasal or naso-tracheal tube, or a tube placed in the oral cavity to the pharynx. Care should be taken with positive pressure ventilations (PPVs) via any of these methods other than tracheal intubation (placed via the oral or nasal cavity), as gases may be forced into the oesophagus and thence the stomach, leading to gastric tympany (Smith et al., 2004).

For animals with suspected or possible respiratory compromise, pre-oxygenate for a few minutes before induction of anaesthesia. The only time this is contraindicated is with induction using inhalational anaesthetic agents administered via a facemask where the patient is stressed by restraint; in this instance, attempts to preoxygenate will likely be counterproductive. Most animals to be induced in a chamber will benefit from oxygen administration prior to the anaesthetic.

BOX 2.1 General hospital requirements for small mammals

- Good-quality food (detailed in species subsections); can ask owners to provide some of usual diet
- Quiet kennel space
- Prey species separated from sight and smell of predator species to reduce stress
- Darkened environment for nocturnal species, such as rats and hamsters
- Species such as rabbits, chinchillas, and guinea pigs that eat hay and use it for bedding will be more settled if good-quality hay is provided, as a food source with a familiar odour (Harcourt-Brown, 2002a)

Analgesia

Analgesia is important for two reasons. Certain analgesic agents will reduce anaesthetic drug requirements, reducing side effects associated. Appropriate and adequate provision of analgesia will also assist during recovery from painful conditions, including surgery. The mu (μ) and kappa (κ) opioid receptors are primarily associated with pain relief in mammals (Paul-Murphy, 2006).

Non-steroidal anti-inflammatory drugs (NSAIDs) inhibit cyclo-oxygenase-1 (COX-1) and COX-2 enzymes. In mammals COX-2 enzymes are involved in inflammation, and both COX-1 and COX-2 are involved in spinal pain transmission (Paul-Murphy, 2006).

FORMULARY

As with other exotic pet species, most anaesthetic agents are not licensed for use in most small mammal pets. However, there are tried and tested protocols for many species, particularly with laboratory animals. Care should be taken with direct extrapolation from laboratory protocols, as these animals will have a higher health specification than pet animals. Some drugs used in mammal anaesthesia, including the narcotic analgesics (for example, fentanyl), may be subject to controls under national legislation. Other agents are discussed in this chapter and later chapters in this section.

Anticholinergics

Anticholinergic drugs are used to protect the heart from vagal inhibition (Harcourt-Brown, 2002a), and are administered to patients with bradycardia. They also reduce bronchial and salivary secretions. However, they may also make secretions more viscous (Bateman et al., 2005) and, therefore, in some cases obstruct narrow airways. Anticholinergics may reduce gastrointestinal motility.

Atropine is the most commonly used anticholinergic in veterinary practice. Forty per cent of rabbits produce atropinesterase, breaking down atropine. Glycopyrrolate is, therefore, used in preference in rabbits. It can also be used in other species, such as rats, guinea pigs and chinchillas.

Medetomidine

The main advantages of this alpha-2-adrenergic agonist in mammals are the good muscle relaxation, the option of subcutaneous or intramuscular administration, the lack of respiratory depression and the option of reversal. This drug causes peripheral vasoconstriction, so mucous membranes have a blue/purple hue (which may appear similar to cyanosis) (Harcourt-Brown, 2002a). Oxygen should always be supplemented when medetomidine is used, as it causes hypoxia (Flecknell, 2000). Medetomidine is often used in combinations to produce more balanced anaesthesia, for example with ketamine. The sedation or anaesthesia resulting varies between species (Nevalainen

Table 2.3: Gastrointestinal prokinetics in rabbits and rodents

DRUG	DOSE (mg/kg)	ROUTE	FREQUENCY	COMMENT
Cisapride	0.5	PO	BID-TID	Not commercially available
Metoclopramide	0.5	PO, SC	BID-TID	–
Ranitidine	2–5	PO, SC	BID	Rabbit

Key: BID = twice daily, PO = orally, SC = subcutaneously, TID = three times daily

(Harcourt–Brown, 2002c; Kounenis et al., 1992; Wiseman and Faulds, 1994)

et al., 1989). Atipamezole can be used to reverse medetomidine and speed recovery.

Gastrointestinal prokinetics

Many herbivore species are susceptible to ileus after anaesthesia, and prokinetics (Table 2.3) are usually administered prophylactically.

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Rabbit anaesthesia

3

INTRODUCTION

The lagomorph most often encountered in practice is the domestic rabbit, *Oryctolagus cuniculi*. A wide range of breeds are kept as pets, ranging from Netherland Dwarfs weighing around 1 kg up to giant breeds, which can weigh 10 kg. The most common breeds presented to veterinary surgeries, such as the Dwarf Lop and Lionhead, weigh 1.8–2.5 kg.

A study into anaesthetic-related death in rabbits showed them to be at increased risk (1.83%) compared to other species (Brodbelt et al., 2005). Animals anaesthetised in poor health or undergoing prolonged procedures were more at risk. Most cases (60%) of mortality occurred post anaesthesia. With this species, more than any other, supportive care will reduce anaesthetic morbidity and mortality (Flecknell, 2006).

ANATOMY AND PHYSIOLOGY

Stress

Rabbits are a prey species and many different factors cause them stress. The effects are varied but ultimately all detrimental to the veterinary patient that in many cases already has underlying pathology. Disease processes, for example dental pathology or pain, will cause stress (Harcourt-Brown and Baker, 2001). Various aspects of husbandry will affect rabbits. These include: inappropriate diet, temperature or companionship; or an inability to behave naturally (Harcourt-Brown, 2002d). In a frightened rabbit, body temperature, heart rate and respiratory rate will be elevated (Donnelly, 2004).

Stress in rabbits leads to release of catecholamines or corticosteroids. Overcrowding has induced cardiomyopathy in laboratory rabbits (Weber and Van der Walt, 1975), and catecholamine release can cause heart failure and death. Sympathetic nervous system stimulation will inhibit gastrointestinal tract activity, reducing motility and digestion. Stress-induced gastric acidity may lead to gastric ulceration. Anorexia associated with the altered carbohydrate metabolism can predispose hepatic disease, initially lipidosis and later liver failure and death. Stress reduces renal blood flow, leading to reduced renal plasma flow and filtration, and decreased urine flow (Kaplan and Smith, 1935). Corticosteroids will also suppress the immune system, predisposing the animal to infectious processes (Harcourt-Brown, 2002d).

Avoidance of the aetiologies of stress in rabbits will reduce complications, not just during anaesthesia but also during hospitalisation. The sections below discuss some important factors to consider when anaesthetising rabbits. It is, therefore, useful to consider sedating or anaesthetising the patient for any stressful procedures. Provision of familiar smells or objects, such as hay or a companion, will provide some security (Harcourt-Brown, 2002d).

Many factors contribute to stress in rabbits, which may lead to problems during and after anaesthesia. Reducing stress is paramount to successful recovery from anaesthesia in this species.

Temperature

Rabbits are very sensitive to heat, and an environmental temperature range of 15–21°C will allow the conscious animal to maintain normal body temperature of 38.5–39.5°C (Batchelor, 1999; Brewer and Cruise, 1994). They should be protected from environmental temperatures below 4°C, and show signs of heat stress above 28°C. Sweating is not effective at heat loss as sweat glands are present only on the lips, and panting does not occur in dehydrated animals (Donnelly, 2004). As thermoregulatory functions are reduced in the anaesthetised animal, supplemental heating will be required during this time and for recovery, but care should also be taken not to overheat patients.

As the only extremity not densely covered in fur, and with a countercurrent arteriovenous shunt, the rabbit's pinnae are important in thermoregulation (Donnelly, 2004). To reduce heat loss during anaesthesia, the ears

can be covered with insulating material such as bubble wrap; conversely, the animal's core temperature can be reduced by cooling the ears, for example with damp towels (Brewer and Cruise, 1994; Cheeke, 1987a).

Cardiovascular system

Normal heart rate can vary from 180 to 250 beats per minute, and is usually higher in smaller rabbits (Brewer and Cruise, 1994; Donnelly, 2004). Blood volume is 55–70 ml/kg (Benson and Paul-Murphy, 1999; Donnelly, 1997).

Cardiac disease is rare, but may include congenital conditions, such as ventricular septal defect or cardiomyopathy (particularly in giant breeds) (Harcourt-Brown, 2002a; Orcutt, 2000). Mitral and tricuspid valvular insufficiencies and valvular endocarditis (Snyder et al., 1976) have been reported. Arteriosclerosis of the aorta and other arteries is reported (Shell and Saunders, 1989). High altitude has also caused pulmonary hypertension (Heath et al., 1990). Heart disease has been associated with various anaesthetics, for example repeated ketamine/xylazine anaesthesia (Marini et al., 1999). An anticholinergic drug, such as glycopyrrolate, may be used to counteract these effects.

Obese rabbits are particularly poor anaesthetic patients, with hypertension and cardiac hypertrophy commonly occurring (Carroll et al., 1996). These patients may also have hyperinsulinaemia, hyperglycaemia and elevated serum triglycerides, and are prone to hepatic lipidosis (Harcourt-Brown, 2002a).

Respiratory system

Anatomy of the rabbit's upper respiratory tract makes visualisation of the larynx and thence intubation a difficult technique. The mouth opening is small, and the oral cavity long and narrow. The tongue is long with a raised fleshy base, the lingual torus. The glottis is small and prone to laryngospasm (Brewer and Cruise, 1994; Cruise and Nathan, 1994). Overweight patients may have a more fleshy oropharynx than other animals, which is more likely to cause upper airway obstruction (Bateman et al., 2005). Rabbits are obligate nasal breathers, and in the normal head position the nasopharynx connects with the larynx (Fig. 3.1).

The thoracic cavity is very small in rabbits in comparison to the abdomen, with correspondingly small lung fields for auscultation (Fig. 3.2). The tidal volume of rabbits is 4–6 ml/kg (Gillett, 1994), with diaphragmatic movements providing most of the impetus for respiratory movement (Harcourt-Brown, 2002a).

Respiratory disease is common in rabbits and any nasal discharge or upper airway inflammation that may occlude breathing is of particular concern when considering anaesthesia. Concomitant lower respiratory tract disease may further compromise respiratory function. The pre-anaesthetic assessment should identify respiratory abnormalities that may cause problems during anaesthesia.

Rabbits respond particularly aversely to the smell of volatile anaesthetic agents such as isoflurane and halothane, and apnoea is common. Bradycardia, hypercapnia and even death can result in some cases (Flecknell et al., 1996). For this reason, pre-medication is given before inhalational agents in rabbits or, more commonly, anaesthesia is induced using injectable agents.

Pasteurella multocida may be found in rabbit nasal cavities without causing disease, but is commonly a secondary invader to primary disease. Predisposing factors, such as poor husbandry leading to immune compromise, will allow replication of the bacteria and resultant systemic pasteurellosis (Harcourt-Brown, 2002c). Many pet rabbits have pneumonia associated with *P. multocida* infection, sometimes with systemic spread to other organs. The possibility of clinical or subclinical respiratory disease should be borne in mind when electing a rabbit anaesthetic protocol.

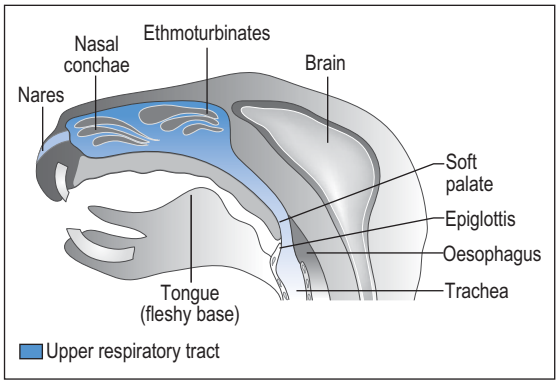


Figure 3.1 • Schematic upper respiratory tract in the rabbit (sagittal section through head). In the normal flexed position of the neck, air from the nares passes to the larynx and trachea. In order to intubate via the oral cavity, the neck must be hyperextended to align the oropharynx with the larynx.

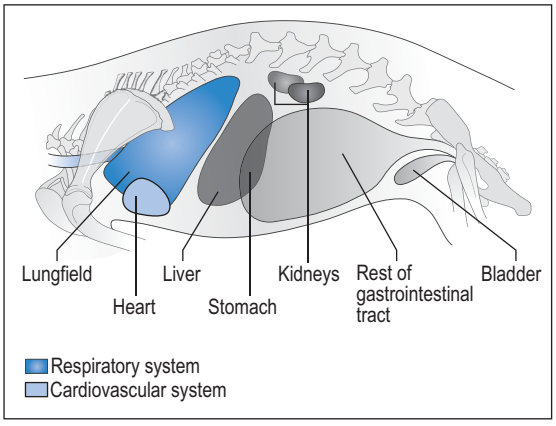


Figure 3.2 • Schematic lateral body view, showing major organs of the rabbit. Note the small size of the lungfield compared to the space occupied by abdominal viscera.

Other infectious agents that may cause respiratory disease in rabbits are *Bordetella bronchiseptica*, *Staphylococcus* sp., *Pseudomonas* sp., *Mycobacterium* sp., Mycoplasma sp. or viruses (Deeb, 2004).

Non-infectious aetiologies of respiratory pathology in rabbits include inflammation due to respiratory irritants or allergens, neoplasia (primary or secondary), cardiovascular disease or trauma (Deeb, 2004). A thorough history to identify predisposing factors, a full clinical examination and investigation, such as imaging techniques, may be required to diagnose the exact aetiology. For the purposes of anaesthesia, it is important to identify that there is a problem and, if possible, to localise it to a particular part of the respiratory tract. The patient should be stabilised with pre-oxygenation prior to choosing an anaesthetic that will cause the least side effects in a compromised animal.

As discussed in the general section, the rabbit thoracic cavity contains small lungs. By comparison, the abdominal viscera are large (Harkness and Wagner, 1995). Problems may arise if positioning allows the large abdominal organs to press against the diaphragm and thence the lungs. Care should be taken to ensure that the rabbit is level or, particularly in dorsal recumbency, at a slight tilt with the thorax raised above the abdomen. Similarly, dorsal recumbency may be associated with more severe and frequent dyspnoea in rabbits (Bateman et al., 2005).

Urinary system

Rabbits drink between 50 and 100 ml of water per kilogram body weight daily, with a total average daily water intake of 120 ml/kg (Cheeke, 1994; Harkness and Wagner, 1995). This volume depends on environmental temperature and water content of food ingested (O'Malley, 2005). Inappetent rabbits may drink excessively, leading to sodium depletion (Brewer and Cruise, 1994; Lebas et al., 1997). Domestic rabbits will drink from water bowls or bottles, and may well have a personal preference. Maintenance fluids are usually administered at 100–150 ml/kg/day, and can either be administered by continuous rate infusion or in three boluses over the day (Table 3.1) (Mader, 2004).

Rabbit urine is normally alkaline (pH 7.6–8.8) with a specific gravity of 1.003–1.036 (Harcourt-Brown, 2002b), and 20–350 ml of urine is produced per kilogram body weight (average 130 ml/kg) daily (Brewer and Cruise, 1994). Assessment of urine parameters in hospitalised rabbits may identify problems that require peri-anaesthetic treatment, such as acid–base imbalances or renal dysfunction.

Many disease processes may affect the rabbit urinary tract. A high-protein diet will increase ammonia levels in rabbit urine (Jenkins, 2004a). Urolithiasis is common in rabbits, and may lead to obstruction and post-renal azotaemia. Urine analysis is useful to assess renal function (Paré and

Table 3.1: Fluid and nutritional support in rabbits

FLUID	ROUTE	DOSE	FREQUENCY	COMMENT
Isotonic crystalloids, lactated Ringer's, dextrose (4%)/ normal saline (0.18%)	IP, IO, IV, SC[1]	Maintenance = 100–150 ml/kg/day	CRI, or divide and administer bolus q6–12 h	Use lactated Ringer's for fluid and electrolyte deficits, dextrose/saline for primary water deficit to support intravascular fluid volume
Glucose 5%	IV, SC[1]	10 ml/kg		Anorexia
Colloids, e.g. hetastarch	IV, IO[2]	5 ml/kg	Repeat if still hypotensive	Hypovolaemic shock. Administer over 5–10 min and assess blood pressure
Liquidised diet: proprietary nutritional support diets (e.g. Critical Care for Herbivores, Oxbow®, Murdock, USA), vegetable baby food, liquidised pellets or vegetables	PO	50 ml/kg/day in total	Divide and give bolus q8h	Anorexic animals Warm food first Use organic, lactose-free baby foods
Blood	IV[3]	10–20 ml/kg, maximum rate 22 ml/kg/h	Can repeat, advise cross-match	Anaemia. Monitor for transfusion reactions. Maximum volume 1% of donor's body weight

Key: CRI = continuous rate infusion, IM = intramuscularly, IO = intraosseously, IV = intravenously, PO = orally, SC = subcutaneously, q8h = every 8 hours

[1] (Harcourt–Brown, 2002a); [2] (Lichtenberger, 2004a); [3] (Lichtenberger, 2004b)

Paul-Murphy, 2004). Dipstick analysis can be used to assess for the presence of protein, glucose, ketones or blood. Haematuria may be due to urinary or reproductive tract pathology. A refractometer is used to measure urine specific gravity and, thus, the concentrating ability of the kidneys. Urine microscopy may also be useful in identifying bacteria, abnormal crystals (calcium carbonate and ammonium magnesium phosphate crystals are found in normal rabbit urine) or cellular composition. Pre-anaesthetic blood biochemistry, radiography or ultrasonography is also useful in cases of suspected renal dysfunction. *Encephalitozoon cuniculi* infection (Flatt and Jackson, 1970) or lead toxicity (Hood et al., 1997) may cause renal pathology and serology or lead assay (respectively) may be useful. If renal disease is identified pre-anaesthetically, fluid therapy should be administered. Anaesthetic agents such as medetomidine may reduce renal circulation and should be avoided in these cases. Safer agents include fentanyl/fluanisone, which does not cause much depression of the circulatory system.

Digestive system

Wild rabbits eat grass and weeds. In captivity they should be given a high-fibre diet of good-quality meadow grass hay, with a concentrate supplement and fresh green vegetables (which are fertiliser- and pesticide-free, and have been washed). A cereal mix allows selective feeding, so concentrates should preferably be extruded pellets. The pellets are usually 15–16% fibre and 16–18% protein. Diets high in protein and low in fibre increase morbidity and mortality, obesity and diarrhoea. Alfalfa hays are high in calcium and protein content, and are useful for growing animals or does that are reproducing or lactating. Correct storage of feed is important to prevent rancidity (particularly if the diet has a high fat content to increase palatability), and to prevent rodent infestation. Water may be offered in a bowl or sipper bottle, depending on what the individual animal is accustomed to. Sipper bottles are preferable in does, which are prone to developing dewlap dermatitis (Brooks, 2004).

Food consumption increases at lower temperatures (Cheeke, 1987b). High temperatures will lead to dehydration through inhibition of drinking and panting, worsened in a low humidity environment (O'Malley, 2005). Intestinal hypomotility will result in decreased colonic absorption of water and electrolytes, leading to dehydration. Therefore, fluid administration is important in cases of hypomotility or ileus in rabbits (Cheeke, 1987c, 1994). Gastrointestinal dysfunction post anaesthesia may result from slow recovery and inappetence (Harcourt-Brown, 2002a).

Any veterinary practice hospitalising rabbits should ensure they provide appropriate food and water in a manner suitable for the individual rabbit. Inappropriate or stale food in a stressful environment will discourage rabbits from eating in the post-anaesthetic period. An unbalanced diet, a sudden change in diet, infections, toxins or administration of certain antibiotics will alter the gastrointestinal microflora, resulting in maldigestion and ileus.

High-fibre diets are necessary to stimulate gut motility and caecotroph production (Cheeke, 1994). It is, therefore, vital that hospitalised rabbits receive adequate fibre in their diet to reduce post-anaesthetic ileus. The autonomic nervous system plays a role in regulation of colonic motility and caecotrophy in the rabbit. Stress (for example, caused by anaesthesia, surgery, illness or diet change) increases adrenaline (epinephrine), which may inhibit gastrointestinal motility and instigate caecal stasis and abnormal caecotrophs (Cheeke, 1987c; Lebas et al., 1997). For these reasons, identification and avoidance of possible stressors (including the provision of analgesia where deemed necessary) will reduce systemic effects. It is necessary to syringe feed high-fibre food to rabbits if they are not self-feeding shortly after anaesthesia (Table 3.1).

Rabbits' body weights will vary throughout the day as the gastrointestinal tract contents vary. Rabbits cannot vomit, so do not generally require fasting before anaesthesia. However, some anaesthetists prefer to fast rabbits for 1–2 h. This will reduce the presence of food in the oral cavity that may be inhaled after induction of anaesthesia, and also reduce gastrointestinal contents that may put pressure on the diaphragm or make abdominal surgery more difficult (Harcourt-Brown, 2002a).

Restoration of the patient's appetite post anaesthesia is important in order to stimulate gastrointestinal motility and to avoid hepatic lipidosis. Nutritional support may be required in the form of syringe feeds, and analgesia may be necessary if pain is present (Harcourt-Brown, 2002a).

Reproductive system

Uterine adenocarcinomas are common in entire does (Baba and von Hamm, 1972; Ingalls et al., 1964; Weisbroth, 1994), with ovariohysterectomy being the treatment of choice. Haematogenous metastatic spread occurs mostly to the lungs and liver, not only affecting the animal's response to anaesthesia but also its prognosis. Thoracic radiographs and abdominal ultrasound may be used to detect these metastases. Various tumours including uterine adenocarcinomas, fibrosarcoma and lymphosarcoma may metastasise to the skin, where tumours may be more readily detected on clinical examination.

If intrauterine haemorrhage has occurred in does with uterine pathology, as evidenced by haematuria or (if haemorrhage is internal) pale mucous membranes, packed cell volume should be assessed before anaesthesia. Fluid therapy and/or a blood transfusion may be required (Table 3.1).

Dystocia is rare in rabbits, but if there is no response to oxytocin, a Caesarean section is indicated (Paré and Paul-Murphy, 2004). Supportive care (warming and fluid administration) should be performed in conjunction with the use of a rapidly reversible anaesthetic protocol. Premedication with a low-dose benzodiazepine should allow mask induction with a gaseous agent or intravenous propofol, before maintenance with a volatile agent.

Nervous system

The initial clinical examination may discover neurological abnormalities, some of which may affect the choice of

anaesthetic protocol. Encephalitozoonosis is a common cause of head tilt in rabbits and may cause simultaneous renal pathology that requires supportive therapy peri-anaesthetically. Similarly, pasteurellosis may result in a head tilt associated with otitis interna or seizures with encephalitis, with the potential for concomitant systemic disease including respiratory infection. Seizures may also be seen with hepatic pathology, including hepatic lipidosis following anorexia. Animals with lead toxicosis will be anaemic and suffer from oxygen deprivation (Deeb and Carpenter, 2004). Metabolic and nutritional imbalances may also lead to neurological abnormalities.

For these higher-risk patients, care should be taken during anaesthesia. An anaesthetic protocol with minimal effects on renal, hepatic or respiratory function should be used, for example pre-medication with midazolam followed by induction and maintenance with isoflurane.

Generalised disease

Some disease processes in rabbits affect more than one body system. Two examples of this are pasteurellosis and lymphoma. Multicentric lymphoma is common in rabbits, with pathology found in many systems, including the upper respiratory tract, abdominal viscera and bone marrow (Huston and Quesenberry, 2004). Clinical signs will vary depending on the location of lesions and are often vague. Thymomas or thymic lymphomas have also been reported (Clippinger et al., 1998; Kostolich and Panciera, 1992; Vernau et al., 1995). Anaesthetic considerations will vary depending on the lesion location and clinical signs associated. The clinician should be aware that more than one organ function might be disrupted.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

As it is one of the larger species covered in the mammals section, with most individuals being used to handling, a full clinical examination should be possible in all rabbit patients.

If cardio-respiratory disease is suspected, the rabbit should be handled gently and for minimal periods to reduce stress. Clinical signs seen in rabbits with cardiovascular disease are primarily tachypnoea or dyspnoea, but more vague signs of lethargy and inappetence may be the only signs noted by the owner. Investigation, including any sedation or anaesthesia required, should be postponed while the animal is stabilised. Sedatives may affect measurements taken by echocardiography (Huston and Quesenberry, 2004).

It is useful to palpate, auscultate and percuss the abdomen before anaesthesia, as individual variation exists, particularly with regard to noises auscultated from gastrointestinal motility. This will enable collection of baseline data for the animal, allowing post-anaesthetic variations to be assessed.

Fluid and nutritional support

Dehydration and electrolyte anomalies may result from a period of anorexia, reduced thirst or specific disease such as diarrhoea or oral discomfort causing hypersalivation (Harcourt-Brown, 2002a). Fluid and electrolyte problems should be identified in the pre-anaesthetic assessment, and attempts made to correct them before administration of drugs (particularly injectable agents) that are likely to have an adverse effect on the circulatory system.

BOX 3.1 Blood loss in rabbits (Jenkins, 2004b)

- Blood volume approximately 57 ml/kg
- Loss of 15–20% total blood volume → massive cholinergic release, tachycardia and intense arterial constriction → redistributes blood away from gastrointestinal tract and skin
- Acute loss of 20–30% total blood volume is critical

BOX 3.2 Checklist for rabbit anaesthesia

- Accurate weight, doses calculated for anaesthetic agent(s)/reversals/emergency drugs
- Supplemental heating, e.g. heat pad
- Intravenous catheter and fluids
- Equipment for intubation – local anaesthetic, laryngoscope, endotracheal tubes
- Anaesthetic machine and circuit
- Monitoring equipment

EQUIPMENT REQUIRED

For rabbits weighing up to 10 kg, an Ayre's T-piece or unmodified Bain's *circuit* is suitable. Paediatric versions are available for small animals (less than 1 kg body weight). Use of paediatric circuits and associated low-volume endotracheal tube connectors will reduce equipment dead space, reducing rebreathing. A mechanical ventilator is useful for performing positive pressure ventilation (PPV) in rabbits (Flecknell, 2006).

Rabbits have a comparatively small laryngeal opening. *Endotracheal tubes* of 2.0–2.5 mm diameter will be suitable for 2.0–2.5 kg animals. Smaller specialist tubes of 1.0–1.5 mm diameter (Cook Veterinary Products (part of Global Veterinary Products Inc.), New Buffalo, MI.) are available for smaller rabbits, and tubes of 5.0–6.0 mm may be required for larger animals. The tubes should be uncuffed. For direct visualisation of the rima glottis during intubation a laryngoscope (with Wisconsin blade of size 0, 1 or 2), otoscope or endoscope is required. A stylet

or introducer formed from a narrow urinary catheter may be useful (Flecknell, 2006).

TECHNIQUES

Routes of administration

Oral

This route is extremely useful for rehydration and nutritional support in rabbits. Dietary fibre is important for gastrointestinal function, and the ability to syringe feed patients with high-fibre supplements is invaluable (for example, Critical Care for Herbivores, Oxbow®, Murdock, USA). Many medications can be administered orally to rabbits. The syringe is inserted to one side of midline, in the gap between incisors and premolars, and a small volume administered at a time to allow swallowing. If the patient is too debilitated to swallow, this technique should be abandoned as aspiration may occur.

Injections

Subcutaneous injections are administered into the dorsal skin over the scapulae or the flank. Large volumes may be given and fluids should be warmed beforehand.

The lumbar or quadriceps muscles are used for *intramuscular* injections. Larger volumes are split between two or more sites to reduce the risk of muscle necrosis.

There are several sites for *intravenous access* in rabbits. The marginal ear veins (Fig. 3.3) are readily accessible in most breeds for venepuncture, both for sampling and catheterisation for administration of fluids and drugs. Alternative sites for venepuncture are the jugular vein (mainly used for phlebotomy, and accessed as in cats with the neck hyperextended), the lateral saphenous vein, the cephalic vein and the mammary vessels.

In the conscious rabbit, it is often useful to apply local anaesthetic cream (for example lidocaine (lignocaine)/prilocaine, EMLA®, AstraZeneca, Södertälje, Sweden) to the skin, in order to reduce the patient's response to needle penetration of the skin. Covering the cream with an occlusive dressing for 15–30 min will allow local anaesthesia to occur prior to catheterisation (Flecknell, 2006); 45–60 min are required for full-skin-thickness anaesthesia (Harcourt-Brown, 2002a). Over-the-needle 23–26-gauge catheters are used to catheterise veins in rabbits.

The lateral or medial edge of the dorsal pinna is clipped and surgically prepared before catheter placement in the *marginal auricular vein*. An assistant raises the vein by applying pressure at the base of the ear (Fig. 3.4). The clinician holds the tip of the pinna with thumb and third finger, and supports the ear margin ventrally with the second finger. The vein is quite superficial, so the catheter should be inserted at an acute angle. Anaesthetics such as medetomidine will cause peripheral vasoconstriction, making intravenous catheterisation more difficult (see Fig. 3.3A). The catheter is usually inserted halfway along the length of the pinna, to allow placement of a bung or connecting device without excess drag on the end of the ear. After advancement of the catheter and removal of the stylet, the catheter can be secured in place with adhesive tape. If the catheter is required for post-anaesthetic use, a light dressing should be used to cover it and prevent the animal removing the catheter. The weight of a catheter with dressing on the ear is uncomfortable for many conscious rabbits, particularly if a fluid line is continuously attached, and may interfere with feeding. The routine use of buster collars to prevent self-removal is contraindicated, as self-feeding is not possible with these collars.

Possible complications with catheterisation of the marginal auricular vein include sloughing of the tips of the pinnae due to chemical phlebitis from infused solutions, mechanical irritation from the catheter or bandage materials (Mader, 2004). In certain breeds with small ears and veins (for example, dwarf breeds), venepuncture is more difficult and may occasionally lead to vasculitis, vascular necrosis and sloughing of the skin or parts of the pinnae (Donnelly, 2004). The central auricular artery should not be catheterised, as complications similarly include damage to the auricular blood supply and subsequent sloughing of part of the pinna (Harcourt-Brown, 2002d).

Catheters in the *lateral saphenous vein* (Fig. 3.5) are better tolerated long-term by rabbits than those in the marginal ear vein. An assistant restrains the rabbit, with the hindlimb held to expose the lateral hock. Skin proximal to the hock is clipped and surgically prepared. The assistant raises the vein by grasping the hindlimb caudal to the stifle. Again, the vein is relatively superficial, but more mobile than the marginal ear vein.

The *cephalic vein* is small and only a short area is accessible in rabbits. It is infrequently used for catheterisation, but can be useful in some cases. Catheterisation will be more difficult in smaller species with a shorter antebrachium (Mader, 2004). Catheter placement is as for other species.

The *jugular vein* can be catheterised, but anaesthesia is necessary for placement of an indwelling catheter.

In very small animals or patients with poor peripheral circulation, *intraosseous* catheterisation into the proximal femur, tibia or humerus can be used to access the circulation and to provide fluids (Ward, 2006). An 18–23-gauge 25–38 mm long hypodermic or intraosseous needle may be used. If required, a stylet of sterile surgical wire can be used within the former needle during insertion to prevent clogging with bone. The animal should be anaesthetised, unless collapsed, and the skin clipped and aseptically prepared. Sterile surgical gloves should be worn. Local anaesthetic is injected near the periosteum. The greater trochanter or tibial crest is palpated and the needle inserted in the same line as the bone, anterograde into the medullary cavity (Fig. 3.6). No resistance should be encountered when a small amount of sterile saline is flushed into the cavity (Mader, 2004). Radiography can be used to check positioning.

Intraperitoneal injections are administered as in other species, usually into the caudal right abdominal quadrant. Fluid absorption is rapid via this route, but there is a risk of viscera perforation.

Intracardiac injections may be required in an emergency situation to administer drugs. The heart is located

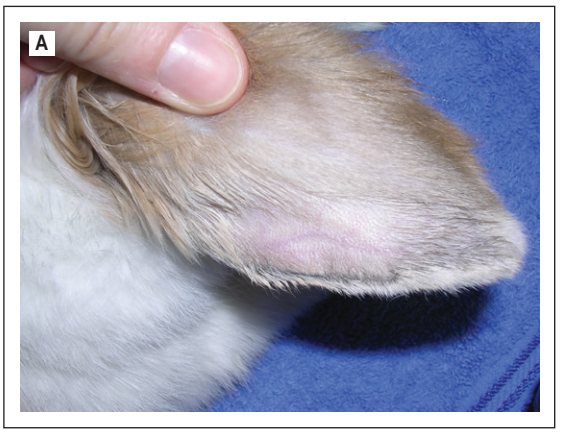




Figure 3.3 • Marginal auricular vein. (A) After medetomidine/ketamine/butorphanol administration. (B) After fentanyl/fluanisone administration.



Figure 3.4 • Intravenous catheter placement in the marginal auricular vein: an assistant raises vein by applying pressure at the base of the ear. The clinician holds the ear at the tip, supporting the cartilage with a finger underneath. The catheter is inserted at a shallow angle into the superficial vein. A light dressing is applied over the catheter.



Figure 3.5 • Catheter in the lateral saphenous vein: the hindlimb is grasped around the caudal stifle to raise the vein for catheterisation. This vein is particularly useful in short-eared breeds such as this Netherland Dwarf where the auricular veins are difficult to access.

between the third to sixth rib spaces near the elbow (Reusch, 2005). Risks include myocardial damage, cardiac tamponade or death.

Intubation

After induction of anaesthesia, when sufficient jaw tone relaxation is attained, rabbits should be intubated. The laryngeal opening is small compared to the size of rabbit, allowing passage of a small uncuffed endotracheal tube, usually a 2.5–3.0 mm for a 2.5 kg rabbit (see Fig. 1.6) (Harcourt-Brown, 2002a). A range of sizes should be prepared for selection depending on the rabbit's size, from 1.5 mm for a 1 kg Netherland dwarf to 5 mm for an 8 kg giant breed. The length of the endotracheal tube should be adjusted if necessary, premeasuring the tube alongside the rabbit so that the connector for the anaesthetic circuit will be at the lips and the tip of the tube within the trachea. It is easier to intubate with a longer endotracheal tube as there is more tube to grasp and manipulate, but the connector should not extend beyond the lips as this increases dead space in the system. A small amount of sterile water-soluble lubricant (for example K-Y Jelly®, Johnson & Johnson, New Brunswick, NJ) may be applied around the tip of the endotracheal tube, ensuring that it does not obstruct the lumen. Care should be taken to avoid traumatising oral and respiratory structures during intubation (Conlon et al., 1990).

Two techniques are commonly used in rabbits, blind intubation and intubation with visualisation of the larynx. In both techniques the neck is hyperextended to align the

Table 3.2: Injection techniques in rabbits

ROUTE	TECHNIQUE	SUGGESTED NEEDLE SIZE AND MAXIMUM VOLUME IN ONE SITE (4 KG ANIMAL)	COMMENT
Intramuscular	Lumbar muscles; quadriceps (cranial thigh)	25-ga, 1 ml	Avoid caudal thigh as risk of damage to sciatic nerve
Intraosseous	Greater trochanter of femur	18–23 gauge needle, 25–38 mm	–
Intraperitoneal	Caudal right quadrant of abdomen, direct needle at 30° angle to skin, withdraw on syringe before inject to ensure viscus has not been pierced	23-ga, 100 ml	Large volumes of fluids can be administered, rapid absorption, warm beforehand; avoid medications which may be irritant
Intravenous	Marginal ear vein, cephalic vein, lateral saphenous vein	22–24 gauge (catheter); 8 ml (bolus), 10 ml/kg (slow infusion)	Marginal ear vein difficult in breeds with short ears, and can be irritating for rabbit to have pinna bandaged Lateral saphenous mobile
Oral	*Syringe:* via diastema *Gavage:* may be aided by use of oral speculum with central hole for passage of lubricated soft feeding tube *Nasogastric tube:* apply local anaesthetic to nares and coat tip of tube with local anaesthetic gel, direct tube caudo-dorsally, radiograph to check placement, secure with butterfly tape sutured to skin on dorsal skull *Oesophagostomy tube:* anaesthesia necessary	Syringe: some formulations require the use of a catheter-tip syringe; Gavage: 13 or 8 Fr tube, 15 ml	Suspension or fluids Premeasure gavage tubes to last rib on left-hand side Care with gavage dosing and nasogastric tube placement, as rabbits may not cough if the tube inadvertently enters trachea Elizabethan collar usually necessary for nasogastric tubes
Subcutaneous	Scruff or flank	23-ga, 30–50 ml	Large volumes of fluids can be administered, warm beforehand, relatively slow absorption

Key: ga = gauge; Fr = French

(Hedenqvist and Hellebrekers, 2003; Mader, 2004; Meredith and Crossley, 2002)

larynx in a straight line from the oropharynx (Fig. 3.7). (In the hyperflexed position, the oropharynx aligns with the oesophagus.) The anaesthetised rabbit is positioned in sternal recumbency with the body in a straight line. An assistant holds the back of the anaesthetised rabbit's head, extending the neck so that the nose–neck–shoulder line is straight and vertical. It may help to lift the rabbit by the head slightly up from the surface.

As the larynx is prone to spasm (Wixson, 1994), local anaesthetic (for example, lidocaine (lignocaine) hydrochloride, Intubeze®, Arnolds) should be applied to it 1–2 min before intubation is attempted. The tongue is grasped and pulled gently out of the mouth, to reduce the obstruction caused by the fleshy base of the tongue, and local anaesthetic sprayed on to the larynx (with or without visualisation; see below). The nose is elevated to allow the local anaesthetic to flow on to the rima glottis, the laryngeal opening.

Blind intubation

In the first technique, the endotracheal tube should be pre-measured, against the side of the rabbit's head, to the level of the larynx. The endotracheal tube is advanced via the oral cavity to the oropharynx. The operator then listens at the connector end of the tube for breath sounds

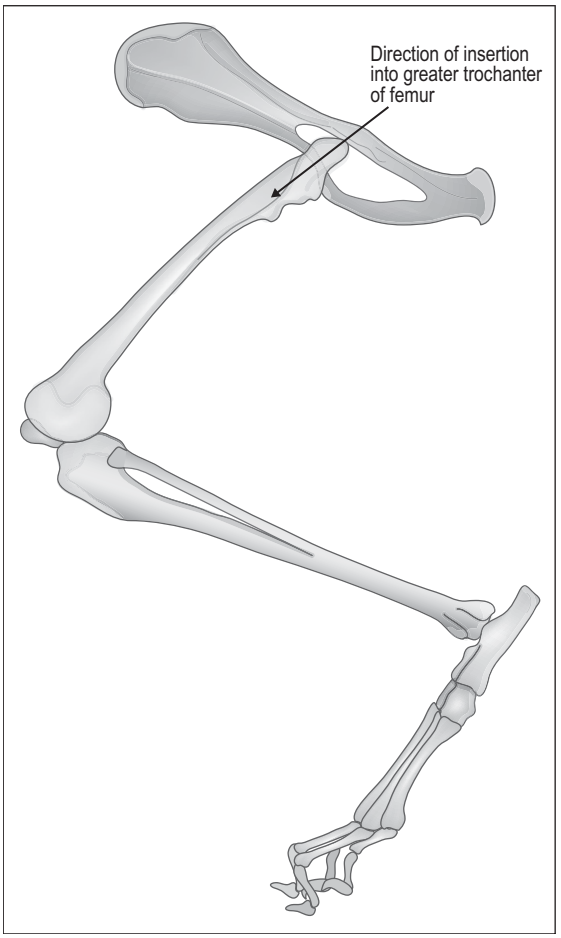


Figure 3.6 • Site for intraosseous catheter placement in the proximal femur of the rabbit.

and advances the tube into the larynx when noise indicates that the larynx is open during inspiration. It can be helpful to observe breathing movements simultaneously or to get the assistant to say when inspiration and expiration occur. If the sounds diminish, the tube has been advanced into the oesophagus and usually some resistance is felt. In this case, the endotracheal tube will be palpable in the neck alongside the trachea. The sounds will be loudest when the tip of the tube is at the level of the rima glottis. As the tube is advanced through the larynx into the trachea, the rabbit may cough (but not in all cases). Breath sounds will still be heard, airflow can be checked by showing movement of a small amount of fur or condensation seen inside clear blue endotracheal tubes or on a glass slide held at the end of the tube, and PPV will cause thoracic movement (Harcourt-Brown, 2002a).

Rabbits do not always cough when a tube or substance enters the trachea.



Figure 3.7 • Hyperextension of the rabbit's neck aligns the oropharynx with the larynx for intubation.

Visualisation of larynx

In the second technique, the larynx is visualised for intubation using a laryngoscope (with a size zero or one straight Wisconsin blade), otoscope or rigid endoscope. The glottis lies deep and caudal in the oro-pharynx (Mader, 2004). The soft palate may initially lie over the rima glottis and can be moved using the tip of the endotracheal tube (Harcourt-Brown, 2002a). In small rabbits, it may be difficult to fit the laryngoscope into the oral cavity without damaging the teeth or soft tissues. If an otoscope with a closed cone is used, a stylet (for example, tubing from a 3–5 French urinary catheter) is first placed into the larynx before removing the otoscope and threading the endotracheal tube over the stylet. If a 1.9 mm semi-rigid endoscope (Needlescope®, Karl Storz, Germany) is available, the endotracheal tube may be guided over this whilst using it to visualise the larynx.

If an initial attempt at intubation is unsuccessful, a smaller endotracheal tube should be used. If either technique is unsuccessful after two or three tries the procedure should be abandoned as the risk of laryngeal trauma and spasm is increased (Wixson, 1994).

Other options for airway maintenance

Laryngeal masks have been used in rabbits with some success. Placement is much easier than intubation and a good

supply of oxygen and anaesthetic gases is applied into the trachea, resulting in reduced environmental contamination compared to facemasks (Smith et al., 2004). However, it is not possible to ensure that the airway is completely protected from material such as fluids in the oral or oesophageal regions, and PPV may lead to gastric dilation. If saliva and gastric contents are aspirated, laryngeal necrosis and pneumonia are likely to occur (Bateman et al., 2005).

Nasal catheters are particularly useful if endotracheal intubation is not possible or if oropharyngeal access is otherwise required for procedures, for example dental treatments. A soft nasogastric tube (size 3–4 French) or small endotracheal tube (1.0–1.5 mm) can be inserted into the nasal cavity to provide oxygen with or without anaesthetic gases (Harcourt-Brown, 2002a). These can be useful even in conscious rabbits requiring supplemental oxygen, although application of local anaesthetic (for example, lidocaine (lignocaine) gel) to the nares and outer surface of the tube will ease their placement and maintenance. In rabbits with incisor tooth disease, the incisor roots may penetrate the nasal airways and preclude placement of a nasal tube.

If endotracheal intubation is not possible via the oral cavity, an alternative is to pass a tube via the nasal cavity into the trachea (Mason, 1997). The muscular nasal fold should be elevated, and the tube directed ventromedially in order to enter the ventral nasal meatus (Flecknell, 2006). The neck should be hyperextended as for the intubation techniques above. This should align the upper airways with the trachea. (If the neck is flexed, a tube passed via the nasal passages will usually pass into the oesophagus.) Remember that rabbits do not always cough when a tube is passed into the trachea. The nasal cavity contains potential pathogens, for example *Pasteurella multocida*, which may be inadvertently introduced into the trachea with this technique (Harcourt-Brown, 2002a).

All anaesthetised rabbits should receive oxygen supplementation. Where none of the above options are possible or appropriate, a *closely fitting facemask* can be used if oral access is not required. If a procedure is being performed on the oral cavity, the end of the anaesthetic circuit or a very small facemask can be held over the nares. Masks are unlikely to provide as good a seal as the other techniques, and so should be used with caution when inhalational anaesthetics are being used, as environmental contamination is likely.

Transtracheal intubation can be performed in an emergency or if upper airway obstruction is present.

Ventilation

It is advantageous to perform IPPV on anaesthetised rabbits, as respiratory depression caused by anaesthesia often reduces tidal volume as well as respiratory rate. The tidal volume of an anaesthetised rabbit is a mere 4–6 ml/kg, although this can be increased to 7–10 ml/kg with PPV. The simplest way of performing PPV is via a mechanical ventilator, but an assistant can perform a similar function using a circuit with valves (Flecknell, 2006). A respiratory rate of 25–50 breaths per minute is appropriate for most patients.

Rabbits have high basal sympathetic tone, and may be sensitive to vagal overstimulation; this may result in arteriolar vasodilation after PPV (Shekerdemian and Bohn, 1999).

PRE-ANAESTHETICS

Drugs that cause sedation in rabbits have several uses. In the first instance, the sedation produced may be sufficient for the procedure to be performed, allowing a rapid return to full consciousness with fewer side effects than a prolonged recovery from general anaesthesia using injectable agents. In the second instance, pre-medication with a sedative will calm the patient, enabling a less stressful anaesthetic induction with other agents, and fewer post-anaesthetic complications. In the third scenario, the premedicant drug may potentiate the other drugs, reducing the doses necessary, and thereby reducing the side effects associated. The fourth use is due to the fact that many premedicant agents have analgesic properties, which is most important if surgery or another painful procedure is to be performed. The final reason for sedating rabbits is that this enables less stressful preoxygenation of the patient during induction, which would not be possible if the rabbit was fully conscious.

Acepromazine may be used on its own as a premedicant or mixed with butorphanol to produce sedation, for example prior to induction using inhalational anaesthetics via a facemask. Acepromazine is vasodilatory, and thence hypotensive. It can be used to produce sedation in rabbits, administered at 0.1 mg/kg subcutaneously or intramuscularly (Heard, 1993). The addition of butorphanol will provide some analgesia (which the acepromazine lacks) along with a mild sedative effect (Harcourt-Brown, 2002a).

The *benzodiazepines* diazepam and midazolam are both routinely used to provide sedation in rabbits, and also cause muscle relaxation. Midazolam is shorter-acting (Flecknell, 1984). Midazolam can be administered intranasally as it is absorbed across mucous membranes (Harcourt-Brown, 2002a). Midazolam affects angiokinesis, reducing the maximum contraction and increasing the speed of relaxation of arteries (Borges and Gomes, 2004).

The benzodiazepines are often used in combination with other agents, injectable or inhalational, for rabbit anaesthesia. Commonly, either of these benzodiazepines can be used to induce anaesthesia after pre-medication with fentanyl/fluanisone (Harcourt-Brown, 2002a).

Medetomidine can be used as premedicant or sedative in rabbits. The resultant peripheral vasoconstriction gives the mucous membranes a blue/purple hue and can make intravenous catheterisation and pulse oximetry more difficult. Medetomidine will also cause hypoxia, and oxygen should always be supplemented when this agent is used (Flecknell, 2000). Mean arterial pressure, heart rate and respiratory rate are usually decreased (Kim et al., 2004); other side effects include hypothermia and diuresis. Advantages of medetomidine include good laryngeal

Table 3.3: Sedation in the rabbit

DRUG	DOSE (MG/KG)	ROUTE	COMMENT
Acepromazine	0.25–1.0[3,4,5]	IM, SC, IV	Mild-to-moderate sedation; duration 4 h Peripheral vasodilation Care in hypovolaemic animals
Acepromazine + butorphanol	0.5 + 0.5[2]	IM, SC	Moderate sedation Peripheral vasodilation, some analgesia Care in hypovolaemic animals.
Diazepam	1–2[2]	IM, IP, SC, IV	Moderate-to-deep sedation; duration 30–180 min Oily preparation can cause tissue damage extravascularly; emulsion preparation safer
Fentanyl/fluanisone (Hypnorm®, Janssen)	0.2–0.3 ml/kg[2]	IM	Mild-to-moderate sedation, moderate to marked analgesia Dose-dependent respiratory depression Reverse fentanyl with buprenorphine or butorphanol
Fentanyl/droperidol (Innovar vet®, Janssen)	0.15–0.44 ml/kg[5]	IM, IV	As for Hypnorm®
Ketamine	25–50[2]	IM, IV	Moderate-to-heavy sedation, some analgesia; duration 1 h (IM), 15–20 min (IV)
Medetomidine	0.1–0.5[2]	IM, SC	Mild-to-profound sedation Peripheral vasoconstriction Respiratory and cardiovascular depression Can reverse with atipamezole
Midazolam	0.5–2[2]	IV, IM, IP	Moderate-to-deep sedation; duration <2 h Sufficient to allow minor procedures or induction with volatile agent
Xylazine	1–5[1]	IM, IV	Mild-to-profound sedation; duration 30–60 min Peripheral vasoconstriction Respiratory and cardiovascular depression Can reverse with yohimbine or atipamezole

Key: IM = intramuscular, IP = intraperitoneal, IV = intravenous, SC = subcutaneous

[1] (Eisele, 1997); [2] (Harcourt–Brown, 2002a); [3] (Heard, 2004); [4] (Jenkins, 1995); [5] (Wixson, 1994)

relaxation (Harcourt-Brown, 2002a) and ease of reversal, of both sedation and side effects, with atipamezole.

In some species, *anticholinergics* are routinely administered as pre-medications. They reduce salivary and bronchial secretions, and reduce bradycardia due to vagal reflexes. In most rabbits this is not required. However, anticholinergic agents should be available for administration in case of bradycardia. Many rabbits possess atropinesterase and glycopyrrolate is the anticholinergic of choice in rabbits. Glycopyrrolate may increase the viscosity of airway secretions and contribute to airway obstruction (Bateman et al., 2005).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Injectable agents

Studies have shown there to be differences in response to various anaesthetic agents between both different rabbit strains (Avsaroglu et al., 2003) and individual rabbits (Aeschbacher, 2001). Certain laboratory rabbit strains

were shown to be resistant to ketamine, medetomidine and propofol; gender differences were also seen. This can make dose selection for an individual difficult. Knowledge of various anaesthetic combinations and application of principles will allow the clinician to vary the protocol if the desired response does not occur.

Many injectable anaesthetic combinations in rabbits will lower blood oxygen saturation levels (Henke et al., 2005). Oxygen should, therefore, always be supplemented either using a facemask or, in preference, an endotracheal tube.

Several anaesthetic agents may affect plasma concentrations of various serum enzymes and biochemical parameters. Combinations of ketamine with xylazine or diazepam may cause increases in alanine aminotransferase, aspartate aminotransferase, blood urea nitrogen, calcium, chloride, cholesterol, creatinine, lactate dehydrogenase, phosphorus, potassium, sodium or triglycerides (Gil et al., 2003; Gil et al., 2004). Values appear to return to control levels within 24 h. Anaesthesia with fentanyl and droperidol did not affect serum values assessed in the study.

When used alone, *ketamine* will cause sedation or can induce anaesthesia. As the eyelids remain open during ketamine anaesthesia, the cornea should be well lubricated (for example, with proprietary liquid paraffin preparations) to prevent damage and ulceration. Moderate respiratory depression is produced. Ketamine causes poor muscle relaxation, and is usually used in combination with other agents for anaesthesia (Harcourt-Brown, 2002a). Ketamine reduces the cerebral vasodilation induced by isoflurane, but not that produced by sevoflurane (Nagase et al., 2003).

A commonly used anaesthetic protocol for surgery in rabbits is the combination of an alpha-2-adrenergic agonist with ketamine. *Xylazine* used on its own will produce moderate sedation, but cardio-respiratory depression is seen and minimal analgesia provided. The xylazine–ketamine combination has significant side effects, including cardiovascular and respiratory depression. Higher doses result in cardiac arrhythmias, and a high mortality rate (Flecknell et al., 1983). Administration of the alpha-2-antagonist atipamezole will reverse the effects of xylazine.

Replacing the xylazine with *medetomidine* in the ketamine combination is associated with a lower incidence of side effects. This combination will also produce surgical anaesthesia (Harcourt-Brown, 2002a; Henke et al., 2005; Nevalainen et al., 1989; Orr et al., 2005). Doses reported for medetomidine and ketamine in laboratory animals are usually higher than those required to produce anaesthesia in pet rabbits. Intramuscular administration produces more rapid induction and recovery compared to subcutaneous injections. Due to the hypoxaemia associated with this combination (Hedenqvist et al., 2001b), supplemental oxygen should be administered. The use of ketamine with medetomidine reduces the change seen in heart rate and respiratory rate when medetomidine is used alone (Kim et al., 2004). Although the heart rate is lowered, no cardiac arrhythmias are produced and only minimal effects are seen on arterial blood pressure with this combination. Blood pressures are higher in rabbits anaesthetised with a combination of medetomidine, ketamine and buprenorphine compared to those where xylazine is used in place of medetomidine (Difilippo et al., 2004).

The anaesthetic period is more prolonged where medetomidine is used in place of xylazine (Difilippo et al., 2004). The period of surgical anaesthesia is also dose-dependent. For example, in one study 15 mg/kg ketamine with 0.25 mg/kg medetomidine produced a mean of 27 min surgical anaesthesia and sleep time of 86 min, compared to 25 mg/kg ketamine with 0.25 mg/kg medetomidine, which produced a mean of 57 min surgical anaesthesia and 103 min sleep time (without atipamezole reversal) (Hedenqvist et al., 2001b).

Medetomidine is regularly reversed with *atipamezole*. Reported doses for atipamezole vary; one study (Kim et al., 2004) recommends atipamezole at equal or double the dose of medetomidine (for example, 0.35 mg/kg medetomidine reversed with 0.35–0.7 mg/kg atipamezole). Five times the dose is given routinely in practice, as this is the same volume of atipamezole (5 mg/ml formulation) to that of medetomidine (1 mg/ml formulation) (Morrisey and Carpenter, 2004).

A combination of ketamine with *diazepam* will decrease respiratory rates, but not heart rates (Gil et al., 2003). Some researchers maintain anaesthesia in rabbits using a continuous rate infusion of either ketamine and fentanyl or propofol, along with isoflurane (Sakamoto et al., 2003). Early work with vitamin C suggests it may also potentiate ketamine anaesthesia in rabbits (Elsa and Ubandawaki, 2005).

Many anaesthetic protocols for rabbits include analgesia, particularly the opioid analgesics, which also have sedative properties. However, respiratory depression is a common side effect; mental depression, hypothermia and bradycardia may also occur. Some opioids, including pethidine, will reduce gastrointestinal motility in rabbits. Most side effects are dose-related, and so can be minimised by using synergistic combinations with other drugs. The addition of butorphanol to ketamine and medetomidine reduces the doses needed of the latter two agents, prolongs anaesthesia and provides analgesia.

Fentanyl is an opioid agonist that acts primarily on μ receptors. The effects of fentanyl are potentiated by *fluanisone* (for example, the fentanyl/fluanisone combination in Hypnorm®, VetaPharma, Leeds, UK or Janssen Pharmaceuticals, Ontario, Canada), a butyrophenone sedative. This combination will provide analgesia for 180 min

BOX 3.3 Commonly used anaesthetics in rabbits

- Fentanyl/fluanisone followed by a benzodiazepine such as midazolam
- Ketamine combinations, for example medetomidine
- Isoflurane (after sedation or induction with other agents)

(Flecknell et al., 1989). Fluanisone also usefully partially antagonises fentanyl's respiratory depressive effects. Fentanyl/fluanisone is vasodilatory (see Fig. 3.3B), hence intravenous catheterisation is facilitated after its administration. As mentioned earlier, anaesthesia may be induced using intravenous midazolam or diazepam after sedation with intramuscular administration of fentanyl/fluanisone.

The fentanyl may be reversed with an opioid agonist/antagonist, such as buprenorphine or butorphanol. These drugs will reverse fentanyl's respiratory depression effects and also continue analgesia for the patient, making them ideal where a painful procedure has been performed. If analgesia is not required post anaesthesia, the pure opioid antagonist naloxone may be used to reverse all of fentanyl's actions. This combination is very safe, but has a prolonged sleep time post anaesthesia, as the sedative effects of fluanisone and the benzodiazepine are still present (Harcourt-Brown, 2002a). This sleep time is more prolonged when higher doses of benzodiazepine are used.

Fentanyl is also produced in a preparation with droperidol (Innovar-Vet®, Jannsen Pharmaceuticals, Ontario, Canada). Bradycardia is commonly seen with this combination (Steffey, 1995).

Medetomidine combined with fentanyl produces anaesthesia. A study on anaesthesia with medetomidine, fentanyl and midazolam showed a high incidence of transient apnoea (Henke et al., 2005). Endotracheal intubation is, therefore, important if this combination is used. This combination has the advantage of the possibility to reverse all components, using atipamezole, butorphanol or buprenorphine, and flumazenil (respectively).

Propofol is a useful induction agent in rabbits. Most patients are pre-medicated, for example with fentanyl/fluanisone (Hypnorm®, Jannsen Pharmaceuticals, Ontario, Canada), before propofol is administered. An intravenous bolus of propofol will rapidly produce sufficient relaxation for intubation. Endotracheal intubation is necessary, as apnoea is common when using this drug and an overdose may cause respiratory arrest (Glen, 1980). Intravenous administration of propofol results in systemic hypotension (Wang et al., 2003). It is not recommended to repeat boluses or to use continuous rate infusions of propofol as the sole anaesthetic agent in rabbits as light anaesthesia only is produced, and hypotension and hypoxaemia are common (Aeschbacher and Webb, 1993b).

Without pre-medication, the ED_{95} for tracheal intubation in rabbits is 6.4 mg/kg (Aeschbacher and Webb, 1993a). If pre-medication is not used, both induction and recovery will be very rapid; however, the higher dose of propofol required to induce anaesthesia will increase the risk of apnoea. Slow administration of the drug, over 30 s, will reduce the risk of apnoea. After intubation, anaesthesia can be maintained via gaseous agents. Propofol is rapidly metabolised, and recovery is smooth and rapid (Harcourt-Brown, 2002a).

Alfaxalone-alphadolone is not recommended in rabbits. The anaphylactic reaction seen in dogs has not been reported in rabbits (Wixson, 1994). This drug will produce a light plane of anaesthesia with muscle relaxation, but no analgesia. Increments can be administered, but high doses can cause respiratory and cardiac arrest (Flecknell, 2000; Harcourt-Brown, 2002a).

Barbiturates, such as thiopental and pentobarbitone, may be used to induce anaesthesia in rabbits, but have a narrow safety margin (Green, 1975). Hypoxia, hypercapnia and acidosis may occur (Flecknell et al., 1983), as may respiratory depression or arrest, and the dose for euthanasia is marginally greater than that for anaesthesia (Wixson, 1994).

Volatile agents

Inhalational agents (including halothane, isoflurane, sevoflurane and desflurane) will induce breath holding and hypoxia in conscious rabbits (Flecknell et al., 1996; Flecknell et al., 1999; Hedenqvist et al., 2001a), and in many under a light plane of anaesthesia (Harcourt-Brown, 2002a). Slow induction with desflurane appears to be best tolerated, but it may take 5 min. Apnoea is usually associated with bradycardia, hypercapnia and hypoxia.

Therefore, in all but the extremely debilitated patient, pre-medication is required before use of inhalation anaesthetic agents. Once the rabbit has been sedated, inhalation agents can be administered via induction chambers or facemasks. Pre-medication will also reduce the volatile agent requirements. For example, in one study (Turner et al., 2006) pre-medication butorphanol reduced MAC_{ISO} from 2.49 to 2.30; this anaesthetic-sparing effect was not seen with the non-steroidal anti-inflammatory drug (NSAID) meloxicam.

If sedation is not used prior to attempted mask or chamber induction, the ensuing breath holding may be fatal. This apnoea is associated with marked bradycardia (Flecknell et al., 1999). Useful sedation agents include acepromazine, fentanyl/fluanisone, and medetomidine (Harcourt-Brown, 2002a). It is also helpful to preoxygenate the rabbit, either via facemask or chamber, before inhalational anaesthetic agents are switched on. This reduces the risk of hypoxia in cases of breath holding (Harcourt-Brown, 2002a).

Isoflurane produces a dose-dependent reduction in respiratory rate and mean arterial pressure in rabbits, but heart rate is not affected (Hayashida et al., 2003).

Anaesthetic maintenance

After induction of anaesthesia is accomplished, either by using injectable agents or by using sedation and inhalation agents, rabbits should, ideally, be intubated. Placement of an endotracheal tube has three benefits: oxygen can be provided, inhalational anaesthetics can be easily and efficiently administered if required for deepening or maintenance of anaesthesia, and PPV can readily be performed (Harcourt-Brown, 2002a). Many rabbits benefit from PPV, particularly when anaesthetised in dorsal recumbency where lungs may become compressed. If it is not possible to intubate the rabbit, for example in very small patients or those undergoing dental treatment where the endotracheal tube will obstruct other procedures, anaesthesia can be maintained using a facemask or intranasal catheter.

Table 3.4: Anaesthetics in the rabbit

DRUG	DOSE (MG/KG)	ROUTE	COMMENT
Atipamezole	0.5[3]	SC, IM	–
Atropine	0.05[6] 0.1–0.2[1]	IM SC, IM	Anticholinergic Many rabbits possess atropinesterase If so, repeat atropine dose or administer glycopyrrolate
Fentanyl/fluanisone (Hypnorm®, Janssen) + midazolam or diazepam	0.2–0.3 ml/kg + 0.5–2 mg/kg[6]	IM + IV (10 min after)	Hypnorm® causes sedation; benzodiazepine then induces anaesthesia Respiratory depression is dose-dependent Reverse fentanyl with buprenorphine or butorphanol Recovery time often correlates with dose of benzodiazepine
Glycopyrrolate	0.01–0.02[1,9]	SC, IV, IM	Anticholinergic[f] – control bradycardia, salivation, or respiratory secretions
Halothane	To effect	Inhal	Pre-medicate before mask or chamber induction
Isoflurane	3–5% 1.5–1.75%[5]	Inhal	Pre-medicate before mask or chamber induction Induction Maintenance
Ketamine	25–50[6]	IM	Painful injection Lack of muscle relaxation means inappropriate for surgical anaesthesia on own
Ketamine + diazepam	10 + 2[4]	IV	–
Ketamine + midazolam	25 + 1[10]	IM	Excellent relaxation; anaesthesia for minor procedures
Ketamine + xylazine	35 + 5[6] 10 + 3[4] 50 + 5[13]	IM IV IM	Can give ketamine when sedated, 10–20 min after xylazine
Medetomidine + fentanyl + midazolam	0.2 + 0.02 + 1.0	IM	Surgical anaesthesia 30 min Transient apnoea common
Medetomidine + ketamine	0.25–0.5 + 25[11,8] 0.25–0.5 + 15[12]	IM	Anaesthesia (loss of ear pinch reflex) (Lower dose of ketamine from study in pet rabbits.)
Medetomidine + ketamine + butorphanol	0.1 + 5 + 0.5	SC, IM	Surgical anaesthesia for 30–40 min (commonly used by author)
Medetomidine + ketamine + buprenorphine	0.5 + 35 + 0.03[2]	IM	Induction of anaesthesia
Naloxone	0.01–0.1[6]	IM, IV, IP	Opioid antagonist
Propofol	3–6[7]	IV	Induce anaesthesia after pre-medication, e.g. 10 min after Hypnorm® IM
Sevoflurane	To effect	Inhal	Pre-medicate before mask or chamber induction
Xylazine + ketamine + buprenorphine	5 + 35 + 0.03[2]	IM	Induction of anaesthesia

Key: Inhal = inhalation, IM = intramuscular, IP = intraperitoneal, IV = intravenous, SC = subcutaneous

[1] (Bateman et al., 2005); [2] (Difilippo et al., 2004); [3] (Flecknell, 2000); [4] (Gil et al., 2004); [5] (Gillett, 1994); [6] (Harcourt–Brown, 2002a); [7] (Heard, 2004); [8] (Hedenqvist et al., 2001b); [9] (Jenkins, 2004b); [10] (Mader, 2004); [11] (Nevalainen et al., 1989); [12] (Orr et al., 2005); [13] (Ypsilantis et al., 2005)

The main advantage of volatile anaesthetic drugs is that they are metabolised much more rapidly than injectable agents, and produce a shorter and smoother recovery from anaesthesia. Halothane may be used in rabbits, but can sensitise the myocardium to catecholamines released during stressful procedures, including anaesthesia. Isoflurane (Lynch, 1986) and sevoflurane (Holzman et al., 1996) depress myocardial contractility less than halothane. Isoflurane is also mainly excreted via the respiratory system, with only 0.2% undergoing hepatic metabolism (Marano et al., 1997). Isoflurane is, therefore, safe for animals with renal or hepatic dysfunction. Induction with isoflurane is rapid, as are adjustments in depth of anaesthesia (Harcourt-Brown, 2002a). Isoflurane (Roth et al., 1996) and halothane (Houghton et al., 1973) provide little or no analgesia.

A light plane of anaesthesia may be suspected if one of several factors is observed. Apnoea or breath holding may occur if the rabbit smells anaesthetic gases. Movement may be seen, particularly if painful procedures are causing stimulation. Vocalisation, often high-pitched, is an alarm response to unpleasant stimuli. This may occur in conjunction with apnoea, and hypoxia or cyanosis may be seen (Harcourt-Brown, 2002a).

The easiest method of deepening the level of anaesthesia is to increase exposure to inhalational agents. Ideally, this is performed via an endotracheal tube, where a short period of positive pressure ventilation without alteration of the concentration of inspired anaesthetic agent may be sufficient. It may be necessary to increase the concentration of inspired agent, especially if the patient has been induced with injectable agents (which have been metabolised) and previously been receiving 100% oxygen. If the rabbit is not intubated, the concentration of inspired agent applied via a facemask should be increased gradually, particularly if the animal is under light anaesthesia, in order to reduce the risk of apnoea in response to the inhalational agent's odour. In situations where gaseous anaesthesia is not possible, injectable agents may be used to 'top up' the anaesthetic. It is useful to have pre-placed intravenous access for this. Great care should be taken in this scenario not to overdose with one particular drug, and it should be remembered that recovery time will be greatly prolonged with top-ups of injectable agents.

Recovery

Anaesthetic gases are switched off and injectable agents reversed if possible. If an endotracheal tube has been placed, it is removed when the swallowing reflex returns (Orr et al., 2005).

Suggested anaesthetic protocols

Fentanyl/fluanisone combinations

The opioid agonist fentanyl is potentiated by the butyrophenone fluanisone. In combination, these drugs produce profound analgesia and deep sedation within 10–20 min of intramuscular injection. These effects are excellent for radiography or for minor procedures such as wound cleaning or dressing changes.

The combination results in vasodilation, allowing ease of phlebotomy or intravenous catheterisation (Fig. 3.3). Analgesia and sedation are provided for 3 h. The dose rate is 0.2–0.3 ml/kg intramuscularly of the Janssen preparation, Hypnorm®, which contains 0.315 mg/ml fentanyl citrate (equivalent to 0.20 mg/ml fentanyl) and 10 mg/ml fluanisone. Subcutaneous administration may produce less sedation, but will be absorbed more slowly.

General anaesthesia can readily be induced after fentanyl/fluanisone using either an intravenous *benzodiazepine* or an inhalational anaesthetic agent. Midazolam (more usually) or diazepam is administered to effect, usually via an intravenous catheter (particularly with the oily preparation of diazepam, which is irritant to tissues when administered extravascularly). The usual dose required is 0.5–2 mg/kg, of either benzodiazepine. Surgical anaesthesia will last for 30–45 min with these regimes, with a sleep time of 4–6 h (Harcourt-Brown, 2002a). The length of sleep time appears to be related to the dose of benzodiazepine administered, with more rapid recovery seen after lower doses.

Alternatively, the rabbit should be sufficiently sedated after fentanyl/fluanisone to allow mask induction. The rabbit is preoxygenated for a few minutes, before anaesthesia is induced using a gradual increase (over 5 min) in *volatile agent* such as isoflurane or sevoflurane. Nitrous oxide can be added (50:50 with inspired oxygen) during induction (Harcourt-Brown, 2002a).

The administration of a mixed opioid agonist/antagonist such as *buprenorphine* or *butorphanol* will reverse the effects of fentanyl, whilst providing further analgesia. Buprenorphine at 0.01–0.05 mg/kg or butorphanol at 0.1–0.5 mg/kg can be administered subcutaneously, intramuscularly or intravenously. Butorphanol will reverse the fentanyl more effectively, but buprenorphine has longer-lasting analgesic actions (approximately 7 h for buprenorphine (Flecknell et al., 1989) compared to 2–4 h for butorphanol) (Harcourt-Brown, 2002a). After reversal, the rabbit may have a sleep time of 1–4 h.

Medetomidine/ketamine combinations

Each of these drugs will produce some degree of sedation, but higher doses of the alpha-2-adrenoceptor agonist medetomidine and dissociative agent ketamine produce side effects. Medetomidine causes bradycardia and respiratory depression. Use of a combination of the drugs enables anaesthesia to be reached using lower doses of each, minimising unwanted effects. The addition of *butorphanol* provides analgesia.

This combination can be administered in one syringe by subcutaneous or intramuscular routes. As relatively large volumes are involved, it is preferable to split the injection into two sites if given intramuscularly. Good restraint is necessary, as the injection may sting, most likely because of the ketamine. Some clinicians prefer to administer the medetomidine subcutaneously, awaiting the sedation 5 min later before administration of ketamine (subcutaneously or intramuscularly) (Flecknell, 2006).

Consciousness is lost 5–10 min after administration of the drugs subcutaneously, or 2–5 min after intramuscular administration (Orr et al., 2005).

As medetomidine leads to hypoxia, supplemental oxygen (via intubation, face mask, or nasal catheter) should always be provided (Hedenqvist et al., 2001b). The depth of anaesthesia may vary between individual animals and supplemental *inhalation anaesthetic agents* are often required for surgical procedures. However, the doses (Table 3.4) should provide sufficient depth of anaesthesia to allow intubation. If inhalational agents are required, the inspired concentration should be gradually increased after a few minutes of preoxygenation, to avoid problems with breath holding (Harcourt-Brown, 2002a).

Nitrous oxide may be used for short periods in rabbits; used as a 50:50 mixture with oxygen it smoothes induction with other inhalation agents. There is a risk of nitrous oxide diffusing into gas-filled spaces such as the caecum when used for prolonged periods. Denitrogenation is required after the use of nitrous oxide, with oxygen being used as the sole carrier gas to the patient for 10 min (Harcourt-Brown, 2002a).

The longevity of surgical anaesthesia varies between animals, but is usually 30–40 min with the medetomidine/ketamine/butorphanol combination. Without butorphanol, surgical anaesthesia is shortened slightly to 20–30 min. The sleep time is usually 90 min, but can be as long as 4 h in some patients (Flecknell, 2006).

The effects of medetomidine, including its analgesic properties, can be reversed using the alpha-2-adrenoceptor antagonist *atipamezole*. As atipamezole is not as long-acting as medetomidine, it should not be administered until a period of 30–40 min has lapsed from medetomidine injection. If atipamezole is administered too soon after medetomidine, resedation may occur (Harcourt-Brown, 2002a). If ketamine has been given with medetomidine, it is advisable to wait 40 min until administering atipamezole, as ketamine alone causes muscle tremors and rigidity (Frey et al., 1996). For a 0.2 mg/kg dose of medetomidine, 1.0 mg/kg of atipamezole is typically administered. The subcutaneous or intramuscular routes may be used to administer atipamezole. Intravenous administration may be used to reverse medetomidine in an emergency, but cardiovascular changes may be rapid and profound.

This anaesthetic combination does not have any long-lasting analgesia effects, with the medetomidine usually being reversed with atipamezole, and butorphanol lasting 2–4 h. Further opioids and/or NSAIDs are routinely used to continue analgesia after surgical procedures performed with this combination.

Propofol

This must be administered intravenously. Most patients will require pre-medication prior to this, unless an intravenous catheter has been previously placed. A low dose of fentanyl/fluanisone (for example 0.15 ml (Kounenis et al., 1992; Wiseman and Faulds, 1994) for a 2 kg rabbit) may be used, causing sedation after approximately 10 min, and vasodilation that assists with catheter placement. Propofol is administered to effect, usually 5–6 mg/kg. Apnoea is common with propofol, and intubation should be performed to facilitate oxygen supply and PPV. Anaesthesia is maintained using volatile agents. Buprenorphine or butorphanol are used to reverse the fentanyl and provide further analgesia. This combination is quite safe for most animals, with rapid metabolism of agents.

Induction with inhalational anaesthetics for neonates or critical patients

Fluid imbalances should be addressed before anaesthesia is induced, or at least an intravenous or intraosseous catheter placed and fluid therapy instigated. Neonates or severely debilitated rabbits are less likely to breath hold during induction with gaseous agents and these may be used to induce and maintain anaesthesia, producing a more rapid recovery than injectable combinations. However, many benefit from pre-medication with analgesia, such as fentanyl in the fentanyl/fluanisone preparation or buprenorphine. All animals should have a period of preoxygenation prior to induction with gases. Nitrous oxide (50:50 in oxygen) may also be administered during induction (Harcourt-Brown, 2002a).

ANAESTHESIA MONITORING

Observations on the patient

Cardiovascular system

The heart rate can be monitored using a bell or in larger animals an oesophageal stethoscope, or very simply by placing a finger on either side of the thoracic cavity near the point of the elbow at the level of the third to sixth rib spaces (Reusch, 2005). The central auricular artery is ideal for monitoring pulse rate and quality. The femoral pulse should also be easily palpable.

Mucous membrane colour is a useful indication of peripheral circulation, but may be altered by anaesthetics such as medetomidine. The normal colour of the nose, lips and tongue is pink. With medetomidine, it will be blue or purple. Any change in colour should alert the anaesthetist to potential problems, such as hypoxia associated with airway obstruction or apnoea. Airway secretions will readily obstruct the airway (IPPV will reduce build-up of respiratory secretions). Neck flexion, even in intubated patients where the endotracheal tube may kink, will also obstruct the airway.

Respiratory system

Monitoring respiratory rate, depth and pattern is paramount to anaesthetic assessment. A respiratory rate of less than 4 breaths per minute is deemed to be severe respiratory depression (Flecknell et al., 1983), and appropriate action should be taken post-haste. Signs of airway obstruction could include a cessation of movement in the reservoir bag, a reduction in oxygen saturation, mucous

membrane becoming cyanotic (blue), a change in respiratory rate or effort, and in severe cases heart rate changes. If signs of airway obstruction are seen, the position of the patient's head and neck should be checked, the oropharynx cleared of fluid or secretions if present, and the tongue pulled forward if the fleshy base may be obstructing the oropharynx (Bateman et al., 2005).

Central nervous system

Depth of anaesthesia is monitored using assessment of various reflexes, which differ from those used in dogs and cats. The most reliable reflex is the toe pinch, leg withdrawal reflex. Rabbits under a surgical plane of anaesthesia will not respond to this stimulus, while those under a light plane may have some tone in their limb muscles and a slow withdrawal. This reflex is more reliable when tested in the hindlimbs than in the forelimbs. Loss of the ear pinch reflex and loss of jaw tone are also useful indicators of surgical anaesthesia (Harcourt-Brown, 2002a).

With most anaesthetics (not ketamine), the nictitans membrane will move over the cornea (Donnelly, 2004). The palpebral reflex is an unreliable assessment of anaesthesia in rabbits. The corneal reflex should not be lost during rabbit anaesthesia, as this occurs only at dangerous depths of anaesthesia. Medetomidine combinations are an exception, where it is routinely lost (Hellebrekers et al., 1997).

> The palpebral reflex is unreliable for monitoring anaesthesia in rabbits.
> The corneal reflex should be maintained throughout anaesthesia.

Anaesthetic monitoring equipment

The heart rate of conscious rabbits is typically between 240 and 280 beats per minute (bpm). These high rates may cause problems with some monitoring equipment. The rate may drop to 120–160 bpm after medetomidine administration (Flecknell, 2000). *Electrocardiography* (ECG) has been used in rabbits (Reusch and Boswood, 2003), but, due to the presence of fur on the ventral surfaces of feet, leads are attached just lateral to the elbows and laterally between stifle and hock. For short procedures, crocodile clips can be applied to skin soaked with spirit to improve contact. The clips can be filed smooth to reduce discomfort (Huston and Quesenberry, 2004; Reusch, 2005). For longer procedures, it is more comfortable for the patient to clip a small area of fur at each of the contact points and to use pads to connect to the ECG monitor.

The central ear artery is useful for placement of *pulse oximeters* to measure oxygen saturation (Herrold et al., 1995). As discussed above, pulse oximetry can be used in rabbits, but reliability of signal production and accuracy of readings are variable. The pulse oximeter can be attached to the rabbit's ear, tongue or digit (Orr et al., 2005). Long-term application of the probe to a rabbit's tongue may cause temporary damage to the lingual muscles.



Figure 3.8 • Indirect blood pressure measurement from the carpal artery in an anaesthetised rabbit.

Arterial *blood pressure* can be measured in anaesthetised rabbits, either directly via the central auricular artery or indirectly using oscillometric limb-cuffs (Ypsilantis et al., 2005). If the central ear artery is used to measure systemic arterial pressure, the blood pressure is lower than in the common carotid artery (by approximately 10 mmHg) (Donnelly, 2004). The direct method is reliable and accurate, but is more technically difficult, and there is a risk of arterial damage and ensuing pinnal necrosis. The indirect method is simpler, but is sufficient to monitor blood pressure routinely in anaesthetised patients. The cuff width is approximately one-third of the circumference of the limb. The cuff is placed around the forelimb just distal to the elbow, with the artery arrow on the cuff dorso-medially over the brachial artery (Fig. 3.8). Alternatively, the cuff is placed over the femoral artery (dorso-medial), proximal to the knee.

End-tidal carbon dioxide can be measured in rabbits. Side-stream samples (Kontron Colormon Plus; Charter Kontron, Milton Keynes, Bucks., UK) add less resistance to the anaesthetic circuit than in-line sampling.

Core body *temperature* is easiest monitoring using a rectal thermometer (Harcourt-Brown, 2002a), but small oesophageal probes can also be used (Sheldrick et al., 1999).

PERI-ANAESTHETIC SUPPORTIVE CARE

Many of the points covered in the mammal introduction section are applicable to rabbits. Particular care should be taken against *hypothermia* with small or very young patients. It is also very important to guard against overheating rabbits, as *hyperthermia* may easily be caused. Signs of hyperthermia include panting (if the animal is sufficiently conscious), seizures and death. It is useful to continue monitoring rectal temperatures during the 'sleep time' post-anaesthesia, ensuring supplemental heating is provided while the animals are recovering, but also to avoid excess heat once the patient is normothermic (see Table 2.1). Electrical heat pads should be removed when

Table 3.5: Commonly used analgesic drugs for rabbits

DRUG	DOSE (mg/kg)	ROUTE	DURATION (H)	COMMENT
Opioids				Analgesic
Butorphanol	0.1–0.5[1]	SC, IM IV	2–4	Both of these agents may cause mild sedation in some rabbits at higher doses, resulting in a slow recovery to normal activity and self-feeding
Buprenorphine	0.01–0.05[1]	SC, IM, IV	6–12	They are also used to reverse fentanyl after Hypnorm® sedation
Fentanyl	0.0074[2]	IV	2–4	Dose-related respiratory depression
Morphine	2–5[3,4,5]	SC, IM	2–3	Affects gastrointestinal motility
Pethidine (meperidine)	5–10[1]	SC, IM		
NSAIDs				Analgesic + anti-inflammatory
Carprofen	4[6]	SC	24	Care in hypotensive animals
	1.5[6]	PO	12	
Flunixin	1.1[1]	SC	12	
Ketoprofen	1–3[6]	SC	12	
Meloxicam	0.2–0.6[1]	PO, SC	24	Palatable oral suspension (Metacam®, Boehringer Ingelheim) well accepted by rabbits

Key: IM = intramuscular, IP = intraperitoneal, IV = intravenous, PO = oral, SC = subcutaneous

[1] (Flecknell, 2000); [2] (Lipman et al., 1997); [3] (Flecknell, 1991); [4] (Heard, 2004); [5] (Jenkins, 1993); [6] (Harcourt–Brown, 2002a)

no longer needed, as rabbits will chew the cables (Harcourt-Brown, 2002a). A digital thermometer measuring room temperature is also a useful monitor.

Administration of *fluids* is useful peri-operatively. They support the circulation, aid metabolism of injectable anaesthetic agents, and can be warmed or cooled to assist maintenance of the rabbit's body temperature. If large boluses of fluids are administered, usually subcutaneously or intraperitoneally, they should be pre-warmed to body temperature (see Table 2.1). If an intravenous catheter has been placed, it is usually left *in situ* with a light dressing until the animal has recovered from anaesthesia, to facilitate administration of emergency medication or fluids if necessary.

As discussed in the general section, provision of a comfortable *environment* post-anaesthesia is important. While good-quality hay provides bedding and food, the rabbit should be placed on a towel or similar surface during recovery, as corneal abrasions may occur in the semi-conscious patient. As soon as the rabbit is sufficiently alert, foodstuffs and water should be provided to encourage a return to normal eating and drinking.

Use of *prokinetics* may not be necessary in all cases. However, prevention of gastrointestinal stasis is much easier than treatment. It is routine to administer at least one dose of a gastrointestinal motility stimulant at the time of anaesthesia, and to continue medication if the rabbit is not producing faeces normally (see Table 2.3). Administration of peri-anaesthetic fluids also reduces the incidence of gastrointestinal disease. If the rabbit is not eating, supplemental feeding should be instigated, usually in the form of syringe feeding (Table 3.1). Placement of a nasogastric or an oesophagostomy tube may be required if anorexia is persistent.

Analgesia

Pain assessment in rabbits is extremely difficult, even more so in the hospital situation where behaviours are affected by other stressors. Individual rabbits will also behave differently when in pain. If the rabbit is showing any signs of discomfort (for example, sitting very still, unresponsive, tooth grinding, inappetent or adopting a crouched position), has a condition likely to be painful in other species, or has been subjected to a painful procedure, analgesia should be administered and continue to be administered until deemed no longer necessary (Table 3.5). Multimodal analgesia is usually employed with administration of both opioid and NSAID drugs.

Local anaesthesia

Topical local anaesthetics, such as proxymetacaine and proparacaine, are commonly used to provide ocular anaesthesia (Mader, 2004). This may be useful, for example, to aid lacrimal cannulation; sedation may be required concomitantly in some rabbits for this procedure. Local anaesthesia may be provided at surgical incision sites using 1% lidocaine (lignocaine) (Hayashida et al., 2003).

Non-steroidal anti-inflammatory drugs

NSAIDs are more effective against somatic or integumentary pain than visceral pain (Jenkins, 1987).

Opioids

Opioids are more beneficial in the alleviation of visceral pain (for example, abdominal surgery) (Harcourt-Brown,

Table 3.6: Emergency drugs in rabbits

DRUG	DOSE (MG/KG)	ROUTE	INDICATION/COMMENT
Adrenaline	0.2[6]	IV, IT	Cardiac arrest (fibrillating or asystole)
Dexamethasone	2[3]	IM, IV	Shock May be ineffective, and may cause severe immune suppression and gastrointestinal ulceration
Diazepam	1[2]	IM, IV, IP	Seizures
Doxapram	2–5[5]	IV, SC	Respiratory stimulant Short duration of effect, may require repeated dosing
Frusemide	0.3–5.0[1,4]	SC, IM, IV, PO	Diuretic
Glycopyrrolate	0.01–0.02[5]	SC	Bradycardia
Lidocaine (lignocaine)	2[7]	IV, IT	Cardiac arrhythmia

Key: ICe = intracoelomic, IM = intramuscular, IO = intraosseous, IP = intraperitoneal, IT = intratracheal, IV = intravenous, SC = subcutaneous

[1] (Allen et al., 1993); [2] (Carpenter, 2005); [3] (Carpenter et al., 1995); [4] (Harrenstien, 1994); [5] (Huerkamp, 1995); [6] (Ramer et al., 1999a); [7] (Ramer et al., 1999b)

2002a). The ultra-short-acting opioid remifentanil has been administered as a continuous rate infusion to provide analgesia in rabbits, producing a dose-dependent decrease in both respiratory rate and heart rate (Hayashida et al., 2003). This agent provides good analgesia and can be reversed with naloxone, but is not routinely used in veterinary practice.

Epidural anaesthesia

The spinal cord in rabbits continues until the sacral vertebrae, the exact endpoint depending on the individual rabbit (Greenaway et al., 2001). As with other species, epidural anaesthesia is useful to provide both intra-operative and postoperative analgesia. If prolonged analgesia is required, a catheter can be inserted for continuous or repeat bolus administration. The location and duration of analgesia produced will depend on the agent and volume used (Dollo et al., 2005). If opioid agonists are used alone, sensory loss occurs. When local anaesthetic agents are used, either alone or concomitantly with opioids, motor and sensory losses are produced; this often results in hindlimb paralysis. If opioids are used only sensory innervation will be lost, and hindlimb function is retained.

Local anaesthetics have been administered epidurally in rabbits to provide analgesia via blockage of sensory and motor nerve fibres (Hughes et al., 1993). Several agents have been administered epidurally in rabbits. High concentrations (5%) of lidocaine (lignocaine) have been shown to have both clinical and histopathological toxicities (Malinovsky et al., 2002). Tetracaine produces similar neurotoxic changes as lidocaine (lignocaine), with bupivacaine less toxic and ropivacaine least toxic (Yamashita et al., 2003). Ropivacaine has been shown to induce dose-dependent spinal anaesthesia without neurotoxicologic lesions. Administering greater volumes of anaesthetic and inappropriate patient positioning, allowing the agent to spread cranially under gravity, are likely to increase 'maldistribution' and associated side effects (Rigler et al., 1991).

Epidural anaesthesia is contraindicated in certain conditions; these include endotoxaemia, meningitis and coagulation abnormalities. Epidural analgesia may protect gut mucosa from injury (Kosugi et al., 2005).

Research into local anaesthetic formulations is ongoing, including mechanisms of bioavailability and clearance (Clément et al., 2004). Various agents can be used to enhance local anaesthetic effects epidurally, for example deoxyaconitine is thought to enhance epidural lidocaine (lignocaine) anaesthesia via κ-opioid receptors (Komodo et al., 2003), or prolong anaesthetic effects (Dollo et al., 2004; Dollo et al., 2005).

EMERGENCY PROCEDURES

If a rabbit does not recover in the expected period of time (which will vary depending on the anaesthetic regime used, the patient's condition pre-anaesthesia and the procedure(s)

performed), the patient should be reassessed. A repeated full clinical examination is warranted, along with a review of investigations carried out so far, and consideration of performing others. There is likely some aspect of ill health that has been missed or not treated sufficiently. Certain problems require drug administration (Table 3.6). Pending a diagnosis, supportive care should continue with oxygenation, fluids and supplemental heat as required.

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4 Rodent anaesthesia

INTRODUCTION

The order Rodentia is subdivided into two suborders (Table 4.1) (Singleton et al., 2004). These divisions are based on various morphological differences. The larger of the suborders is *Sciurognathi*, which includes five families of squirrel-like rodents (including the squirrel family, Sciuridae) and five families of mouse-like rodents. The largest mouse-like rodent family is the Muridae, which includes the pet species of rats, mice, gerbils and hamsters. The *Hystricognathi* suborder has 16 families, with families seen as pets including the Caviidae (cavies), Chinchillidae (which includes the chinchilla), and Octodontidae (octodonts, such as the degu).

Principles of anaesthesia in rodents are discussed at the beginning of this chapter, along with dose rates for anaesthetic agents. Later sections discuss some species differences in anatomy, physiology and pathology that may be relevant when anaesthetising the particular species.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

Inappropriate husbandry may predispose to disease, for example obesity is common in pet rodents. A full history should be obtained, including any known medical conditions. The extent of the clinical examination may be minimal for smaller species, but larger animals, such as guinea pigs and chinchillas, can be fully assessed. All animals should be accurately weighed to ensure correct dosing with drugs and fluids.

Hospitalisation facilities

As for other prey species, a quiet environment away from predator species is conducive to a less stressful

Table 4.1: Taxonomic classification of rodents seen as pets

SUBORDER		FAMILY SEEN AS PETS	SUBFAMILY	EXAMPLE SPECIES
Sciurognathi	Squirrel-like rodents (5 families)	Sciuridae		Chipmunk Prairie dog
	Mouse-like rodents (5 families)	Muridae	Murinae Cricetinae Gerbillinae	Rat, mouse Hamster Gerbil
Hystricognathi	Cavy-like rodents (18 families)	Caviidae Chinchillidae Octodontidae		Cavy, guinea pig Chinchilla Degu

(Singleton et al., 2004)

hospitalisation. Food should be provided that is appropriate for the species in question, along with water in a source recognised by the patient (a sipper bottle or bowl for most rodents). If the practice does not regularly hospitalise rodents, clients can be asked to bring in some of their pet's usual food.

Fluid and nutritional support

Administration of fluids often assists in stabilisation of debilitated patients before anaesthesia. It is possible to administer fluids intravenously, usually accessing the lateral tail vein in rats and mice. As catheterisation and maintenance are difficult, fluids are usually administered as boluses (Table 4.2). Subcutaneous and intraperitoneal administrations are easier, but less rapidly absorbed.

Most fluids administered to rodents will be as a bolus. Large volumes of cool fluids will rapidly lead to hypothermia; all parenteral fluids must, therefore, be warmed to body temperature before administration. A constant-temperature water bath or incubator may be used to warm bags or bottles of fluids before use. The easiest method of checking fluid temperature is to spray a small volume on to your medial wrist (as you would check the water temperature in a baby's bath).

Due to the small total blood volume of rodents, small volumes of blood loss can be significant. Up to 10% of the blood volume can be lost in a healthy animal without any significant effects. However, many pet animals will not be healthy and smaller amounts of blood loss may prove fatal. Blood transfusions from conspecifics may be possible, using intravenous or intraosseous routes for administration.

Table 4.2: Fluid and nutritional support for rodents

FLUID	SPECIES	ROUTE	DOSE (ml/animal)	FREQUENCY	COMMENT
Isotonic crystalloids, Lactated Ringer's, dextrose (4%)/normal saline (0.18%)	Chinchilla	IV, SC	30–60 /day	Divide doses, give q6–12 h	Use lactated Ringer's for fluid and electrolyte deficits, and dextrose/saline for primary water deficit to support intravascular fluid volume. Chipmunk IV/IO doses are for shock therapy
	Chipmunk	SC IP IV IO	2–5 3–5 5–7 5–7		
	Gerbil	IP SC	3–4 2–3		
	Mouse	IP SC	1–3 1–2		
	Rat	IP SC	10–15 5–10		
Colloids	Chipmunk	IV, IO	5–7	–	Chipmunk IV/IO are shock doses
	Gerbil	IV, IO	0.1 (bolus)		
Liquidised diet: proprietary nutritional support diets (Oxbow® Critical Care for Herbivores) Liquidised vegetables or ground pellets Baby food	Chinchilla, chipmunk, guinea pig, hamster, mouse, rat	PO	50 ml/kg/day	Divide, feed q8h	Anorexic animals. Add vitamin C to guinea pig food (10–30 mg/kg/day). Warm food first. Use organic, lactose-free baby foods; vegetarian types for herbivores
Glucose, 5% and 20–50%	All species	SC	10 ml/kg of 4%	Pre-anaesthesia	Use routinely in small rodents, and for pregnancy toxaemia in guinea pigs

Key: IM = intramuscularly, IO = intraosseously, IV = intravenously, PO = orally, SC = subcutaneously, q6h = every 6 hours

(Orr, 2002; Redrobe, 2002, Schoemaker, 2002)

TECHNIQUES

Routes of administration

Oral

Peri-anaesthetic medication may be given orally to conscious patients. For small volumes of palatable medication, the syringe tip is inserted into the patient's mouth just lateral to the incisors. For larger volumes, the gavage technique may be used. To avoid endotracheal administration, the gavage tube diameter should be greater than the tracheal diameter. The patient is restrained and the dosing tube (metal or rubber) passed into the oropharynx and thence the distal oesophagus. A mouth gag should be used if the tube is not metal and may be bitten through by the patient. An inexperienced technician may cause iatrogenic oral, oesophageal or gastric injuries to the animal using the gavage technique (Bihun and Bauck, 2004).

Injections

Anatomic sites for administration of fluids and drugs may vary between species (Table 4.3). Due to the small size of many mammals, there are also maximum recommended

Table 4.3: Routes of drug administration in rodents

ROUTE	SPECIES (maximum volume per site (ml))	COMMENTS
Intracardiac	Hamster, mouse, rat	Palpate apex beat on left thoracic wall between 3rd and 5th ribs, just to left of manubrium
		25-gauge needle
		General anaesthesia required
		Used for emergency administration of drugs
Intramuscular	Chinchilla (0.3)	Quadriceps (see Fig. 4.4); lumbar muscles in larger species. 25–23-gauge needle
	Gerbil (0.05–0.1)	Small muscle mass
	Hamster (0.1)	Injections painful, can cause muscle damage
	Mouse (0.05)	
	Rat (0.3)	
	Guinea pig	
Intraperitoneal	Gerbil (3–4)	Right caudal quadrant of ventral abdomen, animal in dorsal recumbency with injection quadrant tilted up
	Hamster (3–4)	More rapid absorption than subcutaneous route, but some discomfort caused
	Mouse (1–3)	Risk of peritonitis and abdominal adhesions
	Rat (10–15)	Possible in all species, but most often performed under sedation or anaesthesia
Intraosseous	Chinchilla, guinea pig, mouse, rat (as for IV)	Proximal femur, tibia or humerus 26–23-gauge needle in rodents
	Gerbil (0.1 bolus)	Access to vascular system for fluid support and emergency drug therapy
		Useful in severely debilitated animals, also in animals where intravenous access not possible
		Aseptic technique required
		Anaesthesia may be necessary
		Can be maintained for several days
Intravenous	Chinchilla	Technically difficult
	Gerbil (0.2)	Lateral tail vein (not hamster/guinea pig) or lateral saphenous vein; 25-gauge needle; dilate tail vein by warming tail
	Guinea pig Hamster	Administer boluses throughout day, or connect to infusion pump or syringe-driver (e.g. Springfusor®, Go Medical, Australia) to avoid overhydration

(*Continued*)

Table 4.3: (Continued).

ROUTE	SPECIES (maximum volume per site (ml))	COMMENTS
	Mouse (0.2) Rat (0.5)	Cephalic vein possible in larger species, running dorsally and then laterally over tarsal joint; apply tourniquet on proximal antebrachium and use 25- or 27-gauge needle with a heparinised syringe or capillary tube to collect blood
	Chincilla, chipmunk, guinea pig	Jugular vein (general anaesthesia necessary)
	Guinea pig	Anterior vena cava (anaesthesia required)
Oral	All species	Direct administration via syringe or gastric gavage For oral administration, insert syringe just lateral to incisors Small volumes of palatable medication can be mixed with a favourite food For gavage, use soft flexible rubber tubing or a bulb-ended feeding tube Medication in drinking water or food variably accepted and exact dose consumed often not known Conscious animals only Less stressful than SC in some guinea pigs
Subcutaneous	Chinchilla	Scruff or flank
	Degu	25–23-gauge needle
	Gerbil (2–3)	Easiest route; slow absorption
	Guinea pig (25–30)	Do not use flank in gerbils. Guinea pig skin can be thick, especially in males; can be stressful due to discomfort in conscious guinea pigs
	Hamster (3–5)	
	Mouse (2–3)	
	Rat (5–10)	

Key: IV = intravenous, SC = subcutaneous

(Goodman, 2002; Hem et al., 1998; Johnson–Delaney, 2002; Keeble, 2002; Meredith, 2002; Oglesbee, 1995; Orr, 2002; Quesenberry and Carpenter, 2004)



Figure 4.1 • Subcutaneous injection in a guinea pig, *Cavia porcellus*.

volumes for administration to reduce the risk of inadvertent tissue damage.

Subcutaneous injections are generally the easiest to administer, usually in the loose skin of the scruff (Fig. 4.1), and large volumes can be given in this site. However, absorption of drugs from the subcutaneous space is slower than other routes.

Large volumes can similarly be administered *intraperitoneally*. Due to the large blood supply to viscera, absorption is rapid via this route. Intraperitoneal injections are more technically difficult than subcutaneous injections and some substances (including some anaesthetic agents) are irritant when given intraperitoneally.

The animal is restrained in dorsal recumbency with the body tilted so the right caudal abdomen is uppermost (Fig. 4.2). This will allow abdominal viscera to fall away from the injection site and reduce risk of accidental penetration. After cleaning the skin, a small (23–25-gauge) needle is inserted in the right caudal quadrant. For most species, injection into the caudal right quadrant of the abdomen should avoid viscera (Bihun and Bauck, 2004). If aspiration produces any fluid, such as urine or intestinal contents, the needle is withdrawn and the procedure restarted with a fresh needle, syringe and fluids. Placement of the needle through the skin and abdominal musculature causes some discomfort, and is easiest performed in sedated or anaesthetised animals. This also reduces the risk of movement causing inadvertent penetration of viscera.

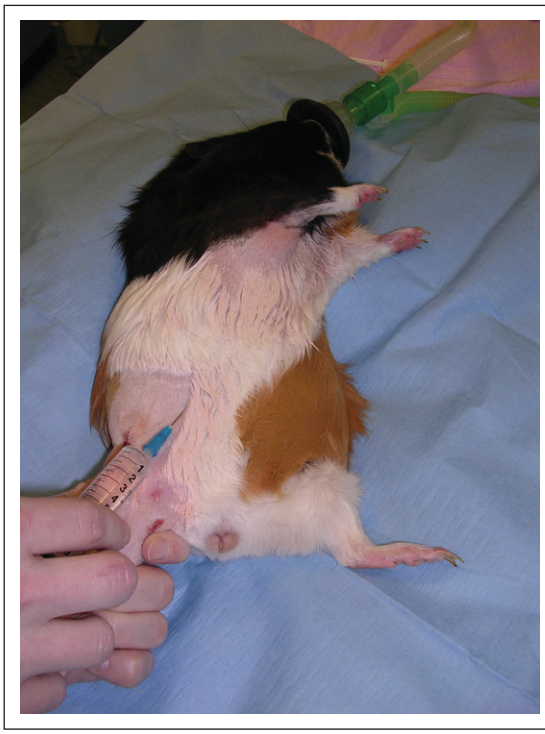


Figure 4.2 • Intraperitoneal injection in a guinea pig, *Cavia porcellus*.

The *intravenous* route is often used in other species for rapid induction of anaesthesia or administration of fluids. This route is difficult to access in most small mammals, especially when conscious. The lateral tail vein is useful, particularly in rats. A hairdryer, incubator (35°C) or warm water (30–35°C) can be used to warm the tail (taking care to avoid heat loss by convection after removal of the tail from the water) to cause peripheral vasodilation. The site should be aseptically prepared. Insulin syringes with 25-gauge needles are selected for ease of injection, although catheterisation is possible using a 24- or 25-gauge bore.

Intraosseous access is an alternative to the intravenous route, although this is only possible in extremely debilitated animals or under general anaesthesia. Analgesia should be administered. In conscious animals, local anaesthetic should be used in the skin and underlying muscle. The proximal femur is commonly used for intraosseous catheter placement (Fig. 4.3) (Bihun and Bauck, 2004). Substances may be administered intraosseously as for intravenous access.

Whilst the intravenous route is often inaccessible for injection of anaesthetic agents, particularly in conscious small animals, the *intramuscular* route is available for rapid drug absorption. The small size of rodent species means that muscle damage is more likely with volumes of agents used. This problem is confounded by the fact that many small mammals have a high metabolic rate and require high doses, and, therefore, larger volumes of drugs compared with other species.



Figure 4.3 • Collapsed degu, *Octodon degus*, with intraosseous catheter in proximal femur for administration of fluids.

The quadriceps group of muscles on the anterior surface of the thigh is most commonly used for intramuscular injections. Alternatively, the gluteal muscles of the hip may be used, avoiding the sciatic nerve in the posterior thigh muscles. If irritant substances are injected near the sciatic nerve, self-trauma to the limb may result in severe damage (Bihun and Bauck, 2004). In larger species, such as the chinchilla, the lumbar muscles may be used for injection of small volumes, but most species have relatively small lumbar musculature.

PRE-ANAESTHETICS

Pre-medication with a sedative (Table 4.4) may ease induction with volatile anaesthetic agents. Certain drugs will also have an anaesthetic-sparing effect, for example morphine will reduce the MAC_{ISO} in rats but meloxicam will not (Santos et al., 2004). Care should be taken when calculating and measuring doses, and should always be based on an accurate body weight.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Volatile agents

The first choice for rodent anaesthesia is complete inhalational anaesthesia (Table 4.5), for example using isoflurane. The main advantages of gaseous anaesthesia are ease of induction and maintenance, including the ability to alter anaesthetic depth rapidly, simultaneous administration of oxygen, wide safety margins with agents currently used, and more rapid recovery compared to injectable agents.

Intubation is not easily possible in these species. If procedures are to be performed on the head or neck, it may not be possible to maintain anaesthesia with gaseous agents without unacceptable leakage into the atmosphere.

Table 4.4: Sedatives and pre-medicants for use in rodents

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Acepromazine	Chinchilla, guinea pig, hamster, mouse, prairie dog, rat	0.5–1.0[3,10]	IM	May induce seizures in gerbils
Atropine	All	0.05–0.1[3]	SC	Some rats possess serum atropinesterase Doses $<$ 0.4 mg/kg reported[2,7]
Diazepam	Gerbil, hamster, mouse, rat Guinea pig	3–5[1] 0.5–3.0[1]	IM	Light sedation, anxiolytic
Fentanyl/droperidol (Innovar–Vet®, Janssen)[1]	Guinea pig Mouse Rat	0.22–0.88 ml/kg 0.2–0.3 ml/kg 0.13–0.16 ml/kg	IM	Sedation Dilute 1:10 to reduce injection site irritation
Fentanyl/fluanisone (Hypnorm®, Jannsen)	Gerbil	0.5–1.0 ml/kg[9]	IM, IP	Moderate sedation Commonly used for minor procedures Reverse fentanyl with buprenorphine or butorphanol
Glycopyrrolate	All	0.01–0.02[6]	SC	Reduce excess oral or respiratory secretions
Ketamine	All	20–40[1,10]	IM	Light sedation at lower dose; heavy sedation at higher dose Marked individual variation Good immobilisation, but poor muscle relaxation Little analgesia (not used commonly)
Ketamine + volatile acepromazine + atropine	Chinchilla	10 + 0.5 + 0.05[5]	IM	Pre-anaesthetic sedation prior to agent induction
Ketamine + midazolam + atropine	Chinchilla	5–15 + 0.5 + 0.05[5,10]	IM	Pre-anaesthetic prior to volatile agent induction
Medetomidine	Gerbil, guinea pig, hamster, mouse, rat	0.1[8]	SC	Light-to-moderate sedation; variable effects in guinea pigs
	Prairie dog	0.5[10]	IM	Hypothermia, cyanosis, hypotension common Medetomidine produces glycosuria and polyuria Reverse with atipamezole

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Midazolam	All	1–2[4]	IM	Light-to-moderate sedation, anxioloytic
Xylazine	Chinchilla	2–10[5]	IM	Light sedation
			IM, IP	Side effects and reversal as for medetomidine (not commonly used)
		2[9]	IP	
	Mouse, rat	10[11]		

Key: IM = intramuscular, IP = intraperitoneal, IV = intravenous, SC = subcutaneous

[1] (Anderson, 1994); [2] (Bennett, 1998); [3] (Drummond, 1985); [4] (Harkness and Wagner, 1995c); [5] (Hoefer and Crossley, 2002); [6] (Huerkamp, 1995); [7] (Ivey and Morrisey, 2000); [8] (Johnson–Delaney, 1999); [9] (Keeble, 2002); [10] (Morrisey and Carpenter, 2004); [11] (Orr, 2002)



Figure 4.4 • Intramuscular injection in the quadriceps muscle in a guinea pig, *Cavia porcellus*. The clinician holds the muscle mass while inserting the needle.

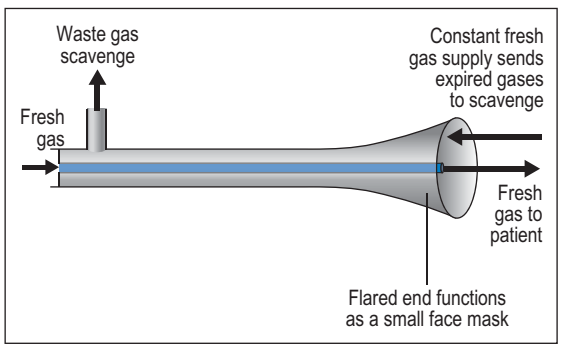


Figure 4.5 • Anaesthetic circuit with a flared nose end for use with small rodents (VetEquip Inc, Pleasanton, CA).

Table 4.5: Suggested concentrations of volatile agents for rodent anaesthesia

AGENT	INDUCTION (%)	MAINTENANCE (%)
Halothane[1,2]	2–5	0.25–3.0
Isoflurane[1,2]	2–5	0.25–4.0
Sevoflurane[3]	To effect (usually higher concentrations required compared to other agents)	To effect

[1] (Anderson, 1994); [2] (Huerkamp, 1995); [3] (Morrisey and Carpenter, 2004)

Most inhalational anaesthetics (the exception being nitrous oxide) do not provide any analgesia and so additional agents should be administered if a painful procedure is to be performed or a painful condition exists.

Rodents may be pre-medicated with one of the protocols above (Table 4.4), but are often induced without pre-medication. Proprietary induction chambers are available, or they can be constructed from any number of different plastic boxes or bottles (see Fig. 1.5). Transparent boxes are ideal, as they allow observation and assessment of the animal during induction.

Preoxygenation of the patient will improve circulatory and tissue oxygen saturation, and is particularly useful in patients with pre-existing cardiac or respiratory disease. The addition of anaesthetic agents to the chamber usually causes some irritation to the eyes and upper airways of the animal, causing the animal to rub its eyes and nose. The use of sedation before induction will reduce the stress this causes to the animal. The anaesthetic agent can either be gradually introduced, starting with a low concentration, or a higher level of agent abruptly introduced. In the first instance, the animal is exposed initially to low concentrations of the agent and anaesthesia is reached more slowly. The second technique causes more initial irritation to the patient, but results in more rapid onset of anaesthesia.

Induction concentrations of 3–4.5% are required with isoflurane and halothane anaesthesia, and 5–6% with sevoflurane (Keeble, 2002; Orr, 2002). Onset of anaesthesia is noted when the righting reflex is lost.

After induction the animal is switched to a close-fitting facemask for administration of anaesthetic, which allows access to the body for procedures to be performed. Several options exist for facemasks in small species, including rodent masks with a clear cone and rubber diaphragm or

circuits with a flared nose end (Fig. 4.5) (both VetEquip Inc, Pleasanton, CA). Impromptu masks can be made from syringe cases attached to the end of the anaesthetic circuit. A significant problem when using masks on rodents is the risk of anaesthetic gas escape from loose-fitting masks into the environment, with attendant risks to staff. Active scavenge is available with some circuits (see Fig. 2.2, Fluovac®, International Market Supply Ltd., Harvard Bioscience Inc., Congleton, UK). Concentrations for maintenance of anaesthesia are lower than those required for induction, typically 1.5–3% for isoflurane and 1–3% for halothane (Orr, 2002). Gerbils appear to require a higher inspired concentration of volatile agents compared to other rodents (Keeble, 2002).

Injectable agents

The second option for anaesthetising rodents is using injectable anaesthesia. A protocol using only injectable agents can be used, or anaesthesia can be 'topped up' or maintained using gaseous agents after induction with injectables. The advantages of injectable anaesthesia are accessibility to the head and neck during anaesthesia, avoidance of environmental contamination with volatile agents, and a lack of requirement for expensive equipment (although it is advisable to provide supplemental oxygen to all anaesthetised animals). The disadvantages with injectable anaesthetics are difficulty of administration, pain on injection or ensuing tissue damage, individual variation in response to anaesthetic drug doses, and an inability to alter anaesthetic depth rapidly. Inter-species differences in response to injection agents exist and genetic variation intra-species has also been shown (Simpson and Johnson, 1996).

> Weigh animals accurately before administration of injectable drugs. Digital scales with 1g increments are necessary for small species.

It is vitally important to have an accurate body weight for the patient before injectable anaesthetics are administered, as it is easy to overdose with these drugs, many of which have narrow safety margins (Table 4.6). It may be necessary to dilute drugs before injection. Most water-soluble compounds will be soluble in sterile physiologic saline (0.9% sodium chloride) or sterile water for injection. A notable exception is the oily preparation of diazepam, which is immiscible in water. Care should be taken when measuring

Table 4.6: Injectable anaesthetics in rodents

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Atipamezole	Guinea pig, mouse, rat	1.0[20]	SC	Reversal of medetomidine Can give IP in mice[2]
Alfaxalone/ alphadolone	Gerbil	80–120[14]	IP	Immobilisation/anaesthesia
	Guinea pig	40[5]	IP	
	Hamster	150[4]	IP	
	Mouse	10–15[4]	IV	
	Rat	10–12[4]	IV	
Fentanyl/droperidol (Innovar–Vet®, Janssen)	Mouse, rat	0.3–0.5 ml/kg[1]	IM	Anaesthesia
Fentanyl/fluanisone (Hypnorm®, Janssen)[19]	Guinea pig	0.5–1.0 ml/kg	IM	Anaesthesia
	Mouse, rat	0.2–0.6 ml/kg	IM, IP	Higher dose required for IP administration
Fentanyl/fluanisone + diazepam[19]	Guinea pig	1 ml/kg + 2.5 mg/kg	IM	Anaesthesia, 45–60 min 120–240 min sleep time
	Mouse	0.4 ml/kg + 5 mg/kg	IP	
	Rat	0.4 ml/kg + 2.5 mg/kg	IP	
Fentanyl/fluanisone/ midazolam*[,20]	Guinea pig	8 ml/kg	IM, IP	As for fentanyl/fluanisone + diazepam
	Mouse	10 ml/kg		
	Rat	2.7 ml/kg		

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Fluamezil + atipamezole + naloxone	Chinchilla	0.1 + 0.5 + 0.05[9]	SC	Reversal for midazolam + medetomidine + fentanyl combination
Ketamine + acepromazine	Chinchilla	40 + 0.5–0.75[15]	IM	5 min to induction, 45–60 min surgical anaesthesia, 2–5 h recovery
	Guinea pig	100 + 5[5]	IP	Light anaesthesia
Ketamine + diazepam	Chinchilla	20–40 + 1–2[10]	IM	Anaesthesia
	Guinea pig	20–30 + 1–2[18]		Diazepam may cause muscle irritation (midazolam preferable)
Ketamine + medetomidine	Chinchilla	0.06 + 5[21]	IM, IP	Anaesthesia; may require volatile agent for surgery
	Guinea pig	40 + 0.5[17]		20–30 min anaesthesia (guinea pig, mouse, rat); 60–120 min (mouse) or 120–240 min (rat) sleep time
	Mouse	50–75 + 1.0[2]		Reverse medetomidine with atipamezole
	Rat	75 + 0.5[17]		
Ketamine + midazolam	Chinchilla, guinea pig, prairie dog	5–15 + 0.5–1.0[11,16]	IM	Light anaesthesia Can also combine ketamine with diazepam for similar effects
Ketamine + xylazine	Chinchilla	40 + 2[11]	IM	2 h surgical anaesthesia (chinchilla)
	Gerbil	50 + 2[1]	IP	May require volatile agent for surgery in some species
	Guinea pig	20–40 + 2[7]	IM	
	Hamster	80 + 5[7]	IM, IP	As for ketamine + medetomidine in mouse/rat, but sleep time (mouse) up to 120 min
	Mouse	50 + 5[7]	IP	
	Rat	75–95 + 5[7]	IM, IP	Xylazine produces glycosuria and polyuria Reverse xylazine with yohimbine
Midazolam + medetomidine + fentanyl	Chinchilla	1.0 + 0.05 + 0.02[9]	IM	Surgical anaesthesia, complete reversal possible (flumazenil, atipamezole, naloxone)
Nalorphine	All	2–5[1]	IV	Narcotic reversal
Naloxone	All	0.01–0.1[12]	SC, IP	Narcotic reversal
Pentobarbitone	Species variability	30–90[1,8]	IP	Narrow safety margin in all species; marginal analgesia Not recommended
Propofol	Mouse	12–26[6]	IV	5 min surgical anaesthesia, 10 min sleep time
	Prairie dog	3–5[16]		
	Rat	7.5–10.0[6]		
Tiletamine/zolazepam (Telazol®, Fort Dodge)	Chinchilla, rat	20–40[10]	IM	Recovery can be prolonged

(*Continued*)

Table 4.6: (Continued).

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Tiletamine/ zolazepam + xylazine	Gerbil	20 + 10[12]	IP	Anaesthesia
	Hamster	30 + 10[7]	IM, IP	
Yohimbine	All	0.5–1.0[7]	IV	Reversal of xylazine

Key: IM = intramuscular, IP = intraperitoneal, IV = intravenous

* One part fentanyl/fluanisone (Hypnorm®, Jannson), two parts sterile water for injection, and one part midazolam (of 5 mg/ml concentration)

[1] (Anderson, 1994); [2] (Cruz et al., 1998); [3] (Eisele, 1007); [4] (Flecknell, 1996a); [5] (Flecknell, 2002); [6] (Glen, 1980); [7] (Harkness, 1993); [8] (Harkness and Wagner, 1995c); [9] (Henke et al., 2004); [10] (Hoefer, 1994); [11] (Hoefer and Crossley, 2002); [12] (Huerkamp, 1995); [13] (Jenkins, 1992); [14] (Keeble, 2002); [15] (Morgan et al., 1981); [16] (Morrisey and Carpenter, 2004); [17] (Nevalainen et al., 1989); [18] (Quesenberry, 1994); [19] (Redrobe, 2001); [20] (Redrobe, 2002); [21] (Röltgen, 2002)

drugs into syringes, as a small error in volume may be a significant error in dose for a small animal. Remember that a one in ten dilution will require one part of anaesthetic agent mixed with nine parts diluent. It should also be noted that the hub in most needles has a relatively large volume, and the use of insulin syringes with the needle directly attached may be more applicable in tiny animals to aid dosing accuracy.

After induction of anaesthesia with injectable agents, oxygen is usually provided via a facemask. If the depth of anaesthesia is insufficient for the procedure to be performed, volatile agents can be added to inspired gases. This is in preference to administration of further injectable anaesthetics, as recovery will be prolonged and the risk of overdose is increased.

Ketamine and *medetomidine* have been used extensively to produce anaesthesia in laboratory rats, with the righting reflex lost within 2–3 min. Both the alpha-adrenergic agents *xylazine* and medetomidine are reported to produce increased diuresis in rats (Waynforth and Flecknell, 1992). The effects appear to be gender-related, with female rats succumbing to deeper anaesthesia compared with similar doses administered to males (Nevalainen et al., 1989). In mice, the effects are reversed, with females requiring higher doses of drugs to produce similar effects (Cruz et al., 1998). As with other anaesthetic agents in mice, this combination produces marked hypothermia (by 4–4.6°C) (Cruz et al., 1998). The effects of ketamine and medetomidine in guinea pigs are variable, with many animals requiring anaesthesia to be topped up with inhalational agents (Nevalainen et al., 1989). Without supplemental oxygen, medetomidine/ketamine combination may produce oxygen saturations as low as 80%.

Recovery

Where volatile agents have been used, recovery is usually rapid when the agent is no longer administered. Some injectable agents may be reversed, but recovery is still more prolonged compared to anaesthesia with volatile agents alone.

In the recovery period, continue to provide *heat* until the patient is moving around. For rats and mice, the initial environmental temperature should be 32°C, reducing to 26–28°C (Orr, 2002). Until the animal is resting in sternal recumbency, it should be turned once or twice hourly to minimise hypostatic pulmonary congestion (Bennett and Mullen, 2004). The patient should be closely monitored until it is able to remain in sternal recumbency. Although a companion may speed an animal's recovery from illness, they should be separated during the immediate post-anaesthesia period as the conscious companion may injure the recovering animal.

The cardiovascular system may be depressed by anaesthetics, as may respiratory movements. To aid *oxygen* saturation, the recovery cage should be oxygen-enriched, if possible, particularly if the patient has respiratory pathology. As discussed above (Pre-anaesthetic assessment and stabilisation section), the cage should also be in a quiet area to minimise stress during recovery.

Appropriate *food* and a *water* source should be provided for the animal. In the recovery period, it can be helpful additionally to supply palatable foods, such as warmed baby food, or soak pellets to increase water consumption (Orr, 2002). Fluids and nutritional support (see Table 4.2) may be required post anaesthesia, particularly if the animal is not observed to be eating and drinking normally within a few hours.

It can be difficult to assess if small animals are eating. Weighing food offered to the animal and the remainder the following day is one technique, but does not readily account for food spilt in the kennel. The easiest method of assessing small patients is to *reweigh* them on a daily basis. Minor fluctuations can be due to urination or defecation, but remember that a few grams weight difference in a 30 g mouse could be a significant 10% weight loss. If any doubt exists over whether an animal is ingesting normal amounts of food and water, supplementation should be given by assist feeding (see Table 4.2).

ANAESTHESIA MONITORING

Observations on the patient

The *respiratory and heart rates* are often too high to physically count in small rodents, and most veterinary surgery ECG machines will not register the high heart rate. It is still useful to observe respiratory rhythm and depth. The use of clear drapes allows better observation of the patient's respiratory movements during anaesthesia.

Rodents have similar *reflexes* to other mammals. The pedal withdrawal reflex is the most useful and is lost at a surgical plane of anaesthesia.

Anaesthetic monitoring equipment

An infant-size bell *stethoscope* can be used to auscultate the heart and lungs. Alternatively a *Doppler* probe may be placed over the heart and used to produce a more easily audible heart rate. *E*CG machines may be of use in larger species, with pads attached to the rodent's feet, but many machines do not register the small electrical deflections in these species. *Pulse oximeters* may be used on the ears or tongue of guinea pigs and chinchillas, or the feet of most rodent species; however, again they may not register a pulse with smaller animals.

A rectal *thermometer* can be used to monitor core body temperature. Digital thermometers are most reliable, and those with external probes are easily used during surgery when drapes cover the animal and surrounding area. The thermometer should be periodically checked to ensure correct positioning.

PERI-ANAESTHETIC SUPPORTIVE CARE

Fasting

As rodents cannot vomit, pre-anaesthetic fasting is not required. In fact, prolonged fasting is contraindicated in these small animals, which have low hepatic glycogen stores and high metabolic rates. The administration of fluids containing dextrose peri-operatively will reduce the risk of hypoglycaemia and dehydration. After induction, the oral cavity (including cheek pouches where present) should be checked for the presence of food material, which may be inhaled during anaesthesia, and cleaned if necessary with cotton-tipped swabs.

Oxygen

Oxygen should be provided to all anaesthetised patients, usually via a small facemask (Fig. 4.6). Some rodents can be intubated (see later), but the technique is difficult. Avoid compromising respiratory function by thoracic compression from equipment or abdominal viscera (Redrobe, 2002).

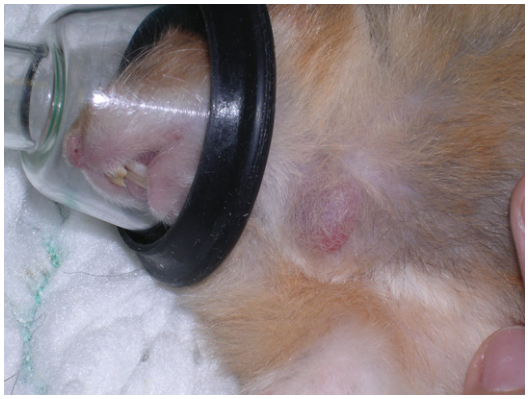


Figure 4.6 • Syrian or golden hamster, *Mesocricetus auratus*, with closely fitting facemask to maintain anaesthesia with volatile agents.

Supplemental heating

Hypothermia is common in anaesthetised rodents, and care should be taken to reduce heat loss and maintain core body temperature. Supplemental heat should be provided during anaesthesia, ensuring heating devices that may cause burns are not in direct contact with the animal. Electric heat pads, heated operating tables, forced warm air blankets (Bair Hugger®, Arizant HealthCare, Eden Prairie, MN), heat lamps, or hot water bottles can be used. Towels and bubble wrap can be used to insulate the animal, including extremities such as feet and tails, and are helpful in minimising heat loss. If skin preparation is required, minimise any fur clipped, use warmed disinfectants, and avoid alcohol-based preparations that may cause heat loss by convection. Hypothermia is not just an immediate problem with reduced metabolic rate, but will lead to slower recoveries as drug metabolism and excretion may be reduced (Robinson et al., 1983).

As with other animals, care should also be taken not to overheat the patient as hyperthermia may occur. Monitor rectal temperatures during anaesthesia and the recovery period. Chinchillas and guinea pigs are particularly susceptible to heat stress, which can be fatal.

Analgesia

Analgesics may be used as sedatives in conjunction with anaesthetic agents. In their own right, they aid recovery from painful conditions and speed the return to normal function in patients. Pre-emptive analgesia is preferred, and multimodal therapy is often indicated.

Pain is a major cause of anorexia in small animals. Analgesia should be administered if a painful condition is suspected or a painful procedure has been performed.

Table 4.7: Analgesics for rodents

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Aspirin (acetylsalicylic acid)	Chinchilla, gerbil, hamster, mouse, rat	100[4,6]	PO	4–8	Great species variability Doses <240 mg/kg q24h reported in gerbil, hamster[10]
Buprenorphine	All Hamster, rat Mouse	0.05–0.1[10] <2.5[3]	SC	6–12	Opioid agonist–antagonist Can be mixed with gelatine for oral administration
Butorphanol	Chinchilla, guinea pig, rat Gerbil, hamster, mouse Prairie dog	0.2–2.0[6,7] 1–5[4,7] 0.1–0.4[2]	SC, IM, IP SC SC, IM	2–4 4 8	Opioid agonist–antagonist
Carprofen	Chinchilla, guinea pig Gerbil, hamster, mouse, rat Prairie dog	4[1,9] 5[8] 1[7]	SC SC PO	24 24 12–24	Non-steroidal anti-inflammatory Care in hypovolaemic or hypotensive animals
Flunixin	Chinchilla Guinea pig, gerbil, hamster, mouse, rat	1–3[5] 2.5[4]	SC	12–24	Non-steroidal anti-inflammatory Care in hypovolaemic or hypotensive animals
Ketoprofen	Chinchilla, guinea pig Gerbil, hamster, rat Prairie dog	1[7] 5[8] 1–3[7]	SC, IM SC SC, IM	12–24	Care in hypovolaemic or hypotensive animals
Meloxicam	Mouse, rat	1–2[1]	SC, PO	12–24	Oral suspension palatable 0.2–0.3 mg/kg q12–24 h used anecdotally in many species
Morphine	Gerbil, guinea pig, hamster, mouse, rat	2–5[4]	SC, IM	2–4	Opioid (narcotic) Not suitable in hamster, as resistant to analgesic effects
Nalbuphine	Gerbil, hamster, mouse, rat Guinea pig	4–8[4] 1–2[4]	IM	3	Opioid agonist–antagonist Used to reverse fentanyl
Oxymorphone	Chinchilla, gerbil, guinea pig, hamster, mouse, rat	0.2–0.5[4]	SC, IM	6–12	Opioid
Pethidine (meperidine)	Chinchilla, gerbil, guinea pig, hamster, mouse, rat	20[3,7]	SC, IM	2–4	Opioid Dose q 6h in chinchilla

Key: IM = intramuscular, IP = intraperitoneal, IV = intravenous, PO = oral, SC = subcutaneous, q6h = every 6 hours

[1] (Flecknell, 2001); [2] (Funk, 2004); [3] (Harkness and Wagner, 1995c); [4] (Heard, 1993); [5] (Hoefer, 1999); [6] (Johnson–Delaney, 1999); [7] (Morrisey and Carpenter, 2004); [8] (Pollock, 2002); [9] (Richardson, 1997); [10] (Smith and Burgmann, 1997)

EMERGENCY DRUGS

Table 4.8: Emergency drugs for use in rodents

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	INDICATION
Adrenaline	Guinea pig	0.003[6]	IV	Cardiac arrest
Atropine	All	0.05–0.1[3]	SC	Bradycardia; excess oral/ respiratory secretions
Dexamethasone	All	4–5[1]	SC, IM, IP, IV	Shock
Diazepam	All	1–5[1]	IM, IV, IP, IO	Seizures
Doxapram	Chinchilla, gerbil, guinea pig, hamster, mouse, rat	2–10[2]	IV, IP	Bradypnoea or respiratory arrest
Furosemide	All	1–10[4]	SC, IM	Pulmonary congestion, oedema
Glycopyrrolate	All	0.01–0.02[5]	SC	Bradycardia

[1] (Carpenter, 2005); [2] (Harkness, 1993); [3] (Harkness and Wagner, 1995c); [4] (Harrestien, 1994); [5] (Huerkamp, 1995); [6] (Laird et al., 1996)

SUBORDER SCIUROGNATHI

Family Muridae (mouse-like rodents)

Introduction

Muridae rodents seen as pets will include those from three subfamilies: Murinae, Cricetinae and Gerbillinae. The Murinae subfamily includes rats (*Rattus norvegicus*) and mice (*Mus musculus*). Cricetinae are hamsters (commonly the Syrian hamster – *Mesocricetus auratus*, but also Russian dwarf hamsters – *Phodopus sungorus* and Chinese hamsters – *Cricetulus griseus*). Gerbillanae are the gerbils (also known as jirds, the most common pet being the Mongolian jird – *Meriones unguiculatus*).

Anatomy and physiology

Temperature

Rodents do not have many sweat glands and cannot pant. Excess heat is lost via the ears and tails, although mice may also salivate to lose heat. All species are susceptible to heat stress (Bihun and Bauck, 2004).

Gastrointestinal system

These species are monogastric, usually herbivorous or omnivorous, and are coprophagic to varying degrees. Coprophagy allows the animals to absorb nutrients including B vitamins. Captive rats, mice, gerbils and hamsters are fed formulated diets (as used in laboratories). These are more balanced than a seed mix and prevent selective feeding (for example, a preference for sunflower seeds from a grain mix). Fatty treats should be avoided as obesity is common in pet animals (Bihun and Bauck, 2004; Orr, 2002). Obesity may compromise cardiopulmonary function during anaesthesia.

Subfamily Murinae (rats and mice)

Temperature

The optimal environmental temperature range for conscious rats is 18–26°C (O'Malley, 2006b; Orr, 2002). Rats have a poor tolerance to heat, having few sweat glands and being unable to pant (Bivin et al., 1979). They reduce their body temperature by radiant heat loss and peripheral vasodilation. The tail is important for thermoregulation and placing it on a warm surface or wrapping it in insulating material will reduce heat loss via convection. Conversely, conscious adult rats are tolerant to cold (Greene, 1962).

Cardiovascular system

The heart contacts the left thoracic wall as the left lung is small and cardiac injections (for emergency access) are possible between the third and fifth ribs (Bivin et al., 1979).

The rat's blood volume is 60 ml/kg. The commonest site for venepuncture in rats and mice is the lateral tail vein, although the smaller lateral saphenous vein and the ventral tail artery are also available (Fallon, 1996). Intravenous access to the lateral tail vein is easier if the tail has been warmed, as peripheral circulation is increased by vasodilation.

Respiratory system

The oval-shaped rat trachea is 3 × 2 mm wide, and bifurcates after 33 mm (Hebel and Stromberg, 1986). It is possible to intubate rats and mice using an inclined support stand for restraint of the anaesthetised animal, holding the mouth open to increase visualisation of the glottis. An otoscope (used as a laryngoscope), local anaesthetic spray, a stylet and small endotracheal tubes (1.22–1.27 mm for a mouse, 14-gauge 5 cm catheter for a rat) are then utilised as for intubating larger species (Kastl et al., 2004). Transillumination of the trachea may aid visualisation of the larynx (Remie et al., 1990). This procedure is only routinely performed in laboratories and not usually in veterinary practice. Small endotracheal tubes may readily block with airway secretions, although this risk may be reduced using positive pressure ventilation (PPV) (preferably via a mechanical ventilator).

Respiratory disease is prevalent in pet rat and mouse populations (Donnelly, 2004b). Clinical signs are usually obvious on examination, including dyspnoea, respiratory noise, sneezing, nasal discharge and stress-related chromodacryorrhoea (red oculonasal discharge of porphyrins from the Harderian glands). Upper respiratory tract irritation to ammonia or dusty bedding may cause mild signs or predispose to infections. Pneumonia is common in rats. Infectious aetiologies usually cause more severe signs, with *Mycoplasma pulmonis* being the most common agent in chronic respiratory disease in rats. Often other agents are involved concomitantly, such as *Streptococcus pneumoniae*, *Corynebacterium kutscheri*, Sendai virus and cilia-associated respiratory (CAR) bacillus (Orr, 2002).

Sialodacryoadenitis virus is a coronavirus. Initially, infection causes a rhinitis, before disease progresses to involve the salivary and lacrimal glands. The upper respiratory tract lumen is narrowed due to inflammation, compromising the patient's breathing, and anaesthetic deaths are common (Donnelly, 2004b).

Digestive system

As with other rodents, access to the airways is made more difficult by a long narrow oral cavity and the caudal base of the tongue is raised into the lingual torus (Bivin et al., 1979). The stomach has an acute angle at the lesser curvature that precludes vomition, and so fasting is not required before anaesthesia.

Cedar bedding affects microsomal oxidative liver enzymes in rats and mice. Clinical signs have not been associated with these changes, but they may affect drug metabolism (Weichbrod et al., 1988).

Urinary system

Rats concentrate urine well, and normal urine output is 15–30 ml daily. Proteinuria may be normal (Bivin et al., 1979). Polydipsia and marked proteinuria (10 mg/l) may suggest chronic progressive nephropathy, which is common in aged rats (Orr, 2002). Assessment of blood urea nitrogen may be required to investigate suspected renal disease.

Special senses

Rats communicate at frequencies outwith the range of human hearing and can hear ultrasonic frequencies up to 60–80 kHz (Koolhaas, 1999). They are sensitive to high-pitched and ultrasound noises from equipment such as computers (Gamble, 1976), but studies show that the cardiovascular system is not affected by ultrasound noise (20–40 kHz) as it is by audible noise (Burwell and Baldwin, 2006). A quiet environment is thus important to reduce autonomic changes in hospitalised animals.

The olfactory system in rats is particularly well developed (Sharp and LaRegina, 1998). Care should be taken to avoid inappropriate smells (for example, from other animals, including bedding from unfamiliar conspecifics) that may stress the patient in the hospital environment.

Subfamily Gerbillinae (gerbils)

Temperature

Gerbils have adapted to great variations in environmental temperature, from −40°C in winter to over 50°C in summer in their wild desert habitat (Keeble, 2002). Relative humidity higher than 50% will cause them stress (Donnelly, 2004b).

Cardiovascular system

The total blood volume of a gerbil is approximately 70 ml/kg (Keeble, 2002). Venepuncture sites include the lateral tail vein and saphenous vein (Hem et al., 1998).

Digestive system

Wild gerbils eat coarse grasses, roots, seeds and occasional invertebrates (Agren et al., 1989). In captivity they are mainly fed rodent mix, and fresh fruit and vegetables. Some will eat hay. Occasional treats may be given. Water is provided in a bottle (Keeble, 2002).

Tyzzer's disease, caused by *Clostridium piliforme*, can cause fatal diarrhoea along with hepatic lesions (Motzel and Gibson, 1990). Many gerbils become obese when fed on captive rat or mouse mixed diets, with some developing diabetes (Donnelly, 2004b).

Respiratory system

Gerbils can be intubated, but the technique requires specialist equipment and is not routinely performed in practice. Tracheotomy may be performed, or specialised laryngoscopes and endotracheal tubes used (Huerkamp,

1995). Intravenous catheters (without the stylet) can be used, but are easily occluded by respiratory secretions as in other small mammals (Antinoff, 1999).

Urinary system

As desert species, gerbils are highly adapted to conserving water. They produce small volumes of concentrated urine and only require low volumes of water intake. Urine is normally alkaline, and may contain protein, glucose and acetone in low levels (Keeble, 2002). Polydipsia/polyuria and weight loss may be found with chronic interstitial nephritis, which is common in ageing gerbils (Donnelly, 2004b).

Endocrine system

Diabetes mellitus may occur in obese gerbils (Laber-Laird, 1996). These animals will have problems with glucose metabolism and are susceptible to hepatic lipidosis if diet is rapidly altered (Keeble, 2002).

Nervous system

Certain genetic lines of gerbils commonly have spontaneous epileptiform seizures (Laming et al., 1989). A change in environment or handling may stimulate a seizure (Keeble, 2002). With these individuals, minimising stimulation (including handling and loud noises) can reduce seizures. Although the zoonotic virus lymphocytic choriomeningitis is often asymptomatic in rodents, it may cause seizures in gerbils (Harkness and Wagner, 1995d).

A head tilt may be due to otitis media or interna related to bacterial respiratory infections (Keeble, 2002).

Fat-tailed gerbil or fat-tailed jird (Duprasi)

These rodents are similar to the Mongolian gerbil, but belong to a different group in the Gerbillinae subfamily. However, the fat-tailed gerbil, *Pachyuromys duprasi*, is more insectivorous, eating some fruit (Johnson-Delaney, 2002; Kingdon, 1997). Captive diets are similar to that of African pygmy hedgehogs, except feed can be *ad libitum* unless obesity occurs. Captive animals often suffer from obesity, particularly if fed on grain-based diets.

Subfamily Cricetinae (hamsters)

The most common pet hamster is the Syrian or golden hamster (*Mesocricetus auratus*). Dwarf hamsters, such as the Roborovski (*Phodopus roborovskii*) and Djungarian (*Phodopus sungorus*), may also be seen.

Temperature

The optimum environmental temperature range for hamsters is 20–24°C (Bivin et al., 1987). Below 10°C, hamsters will hibernate (Goodman, 2002). They have a high metabolic rate, and are prone to heat and fluid loss. They are particularly stressed in hot and humid environments (Bihun and Bauck, 2004), and temperatures and relative humidity should be monitored during hospitalisation using a digital thermometer and hygrometer. During recovery from anaesthesia, the environmental temperature for a hamster should be 35–37°C (Goodman, 2002).

Cardiovascular system

The midline heart in hamsters contacts the thoracic wall between the third and fifth ribs. The normal heart rate is 250–500 beats per minute. Blood volume in Syrian hamsters (*Mesocricetus auratus*) is 78 ml/kg (Bivin et al., 1987). Atrial thrombosis (Hubbard and Schmidt, 1987) and congestive heart failure caused by cardiomyopathy have been reported in hamsters (Donnelly, 2004b).

Venepuncture is difficult in hamsters and sedation is required. Sites for injection are restricted to the lateral saphenous, jugular or cephalic veins, although the anterior vena cava or cardiac puncture are options for emergency administration of medication (Goodman, 2002; Whittaker, 1999). Hamsters have a large amount of loose dorsal skin interscapularly, enabling easy injection of large volumes of fluids (O'Malley, 2006a). However, fluids are slowly absorbed from this large potential space.

Respiratory system

Normal respiratory rate is 30–32 breaths per minute (Bivin et al., 1987). Pneumonia is common in pet hamsters (Donnelly, 2004b). *Streptococcus* spp. causing bacterial pneumonia may originate from their human carers. A poorer prognosis should be given for animals with pneumonia with concomitant purulent rhinitis and ocular discharge (Kuntze, 1992).

Digestive system

Hamsters are mainly herbivorous, normally eating seeds, shoots and root vegetables, but also consuming leaves and flowers (Feaver and Shibin, 2004). This species feeds in short (5 min) bursts with 2-h fasts between (Bivin et al., 1987). Food intake is 5–7 g daily (Newcomer et al., 1987).

The base of the tongue is muscular (Bivin et al., 1987). Vomiting is impossible, as the lesser curvature of the stomach is very short, with the cardia near the pylorus (Hoover et al., 1969; Lipman and Foltz, 1996). Fasting is not required before anaesthesia, but the large cheek pouches should be emptied on induction to reduce the risk of aspiration of stored food material.

Urinary system

Normal hamster urine may vary widely in pH, from 5.1 to 8.4. They drink 10 ml water per 100 g bodyweight daily (Goodman, 2002; Newcomer et al., 1987). Urine production is usually 7 ml per day (Syrian hamster), but this may increase 10-fold in diabetic individuals (Harkness and Wagner, 1995b). As is the case with rats, proteinuria may be normal (O'Malley, 2006a).

Special senses

These nocturnal animals have a well-developed sense of smell and acute hearing; including an ability to hear at ultrasonic levels (Feaver and Shibin, 2004). Efforts should be made to reduce stress on hospitalised individuals by minimising strong odours and loud noises.

Pre-anaesthetic assessment and stabilisation

History and clinical examination

Most mouse-like rodents are nocturnal, although to varying degrees. For example, rats are more nocturnal than mice. They should be woken gently to avoid evoking a bite response. The small size of many patients limits clinical examination and history taking may be more important in identifying potential causes or predisposing factors for disease.

Observation of the patient before handling may identify cardio-respiratory disease, for example lethargy, noisy respiration or dyspnoea. It is useful to observe respiratory movements and measure respiratory rate (not possible in smaller species as often too rapid to count) at rest, as the stress of handling may significantly alter the breathing pattern. If dyspnoea is noted, handling should be minimised to avoid stress and possible mortality.

Pre-existing disease may well compromise cardiopulmonary function during anaesthesia. The identification of disease in these animals may in itself not be straightforward, although careful history taking and clinical examination may reveal abnormalities. In an ideal situation, urine analysis, blood biochemistry and haematology should be performed prior to anaesthesia. Dipstick analysis and specific gravity can be performed on quite small urine samples collected on to a clean non-absorbent kennel liner (for example, an upturned incontinence pad). However, in most patients even venepuncture will require anaesthesia. Familiarity with the species in question, including knowledge of good husbandry practices, normal body condition and behaviour, aids in clinical assessment of the patient pre-anaesthesia.

An accurate weight (to the nearest gram in species as small as mice) is essential before the administration of medications, as accidental overdosage is easy.

Induction and maintenance of anaesthesia

The small size of Muridae species precludes some anaesthetic techniques routinely used in larger species and makes others technically difficult.

Intubation is not routinely performed in pet rodents and intravenous access may not be possible. Injectable agents can be administered via the subcutaneous, intramuscular or intraperitoneal routes. The injectable anaesthetic drugs cannot, therefore, be given gradually to effect, and much individual animal variation in response to anaesthetics exists. In most cases, volatile anaesthetics are used for induction and then also maintenance of anaesthesia in rodents (Johnson-Delaney, 2002). A chamber is used to induce anaesthesia, transferring to a small facemask or nose cone for maintenance. Sevoflurane may be used, but haloalkenes produced by contact with carbon dioxide absorbents are reported to cause nephrotoxicity in rats (Patel and Goa, 1996). There are instances where injectable agents are required, often in conjunction with volatile agents.

Doses appropriate for hamsters are used in fat-tailed gerbils.

Family Sciuridae (squirrels)

This family includes the chipmunk (*Tamias sibericus*) and prairie dog (*Cynomys ludovicianus*).

Chipmunks

Chipmunks are omnivorous, with their wild diet mainly comprising seeds, buds, leaves and flowers. The diet in captivity is commercial dry mixes, along with fresh and dried fruit, vegetables and nuts. Some dog biscuits and animal protein (mealworms, cooked meat, hard-boiled eggs and day-old chicks) may be offered (Meredith, 2002). Water is usually provided in a sipper bottle.

This species is less commonly seen as pets than other rodents. Chipmunks are very susceptible to stress, including noise, overcrowding and being caged in a confined space. Prolonged exposure to the electromagnetic and ultrasonic radiation from televisions can result in death. After transportation or other stressful event, a chipmunk may be subdued for 24 h (Meredith, 2002).

As many pet chipmunks are not used to handling and become stressed when caught, general anaesthesia is often required for clinical examination. Gaseous anaesthesia is usually easiest, as minimal handling is required prior to placing the animal in an induction chamber.

Few drug doses are published for this species, but many clinicians extrapolate from rat doses. Chipmunks require 75–100 ml/kg of fluid daily for maintenance (Meredith, 2002).

Prairie dogs

The black-tailed prairie dog (*Cynomys ludovicianus*) is uncommonly seen in the UK, but some pet animals are present in the USA. These animals like to burrow, so deep substrate, such as shredded paper, should be provided during hospitalisation. Prairie dogs may transmit a variety of zoonotic infections, including *Yersinia pestis* and *Salmonella* (Funk, 2004).

Temperature

The optimum environmental temperature for prairie dogs is 20–22°C, with relative humidity between 30 and 70% (Johnson-Delaney, 1996; Lightfoot, 1999). Dormancy is induced at temperatures below 16°C.

Cardiovascular system

Animals over 3 years of age frequently develop dilated cardiomyopathy (Lightfoot, 1999, 2000).

Venepuncture may be possible in conscious prairie dogs using the lateral or medial saphenous vein, cephalic vein

or jugular vein. The cranial vena cava may be accessed in anaesthetised animals.

Respiratory system

Various environmental conditions predispose to respiratory problems in prairie dogs, including poor ventilation, high humidity and excess dust. Obese animals are often dyspnoeic (Funk, 2004). Sinusitis, rhinitis, cardiomyopathy and dental disease (such as infection or neoplasia) may cause upper respiratory tract problems. Many wild-caught animals will have pulmonary mites (*Pneumocoptes penrosei*), which may lead to dyspnoea by occluding the nasal passages. Pneumonia may be caused by bacterial (for example, *Pasteurella multocida*), fungal (for example, *Aspergillus* sp.) and mycoplasma (Johnson-Delaney, 2002) infections. Pre-anaesthetic stabilisation of dyspnoeic animals may require oxygen therapy, nebulisation, appropriate antimicrobials and bronchodilation.

Digestive and urinary systems

Prairie dogs in the wild graze on grasses, and also on leaves, herbs and flowering plants. They will occasionally take some invertebrates and, rarely, carrion (Funk, 2004). As hindgut fermenters, prairie dogs require adequate roughage in their diet, so captive animals should receive unlimited grass hay. Juveniles can also receive pelleted chows and alfalfa *ad libitum*. Pellets should be limited if the animal becomes obese or when they reach adulthood. Treats include small amounts of fresh greens. Obesity is common in captivity (Johnson-Delaney, 2002).

Both hepatic and renal neoplasias have been reported in prairie dogs (Griner, 1983; Tell, 1995; Woolf et al., 1982).

Anaesthesia of Sciuridae

Induction and maintenance of anaesthesia are usually performed with isoflurane. A chamber or facemask is used for induction. Usually volatile anaesthetics are administered via a facemask to maintain anaesthesia. Endotracheal intubation is possible and is performed similarly to rabbits using either a blind technique or visualised with a laryngoscope. Endotracheal tubes of 2.0–2.5 mm can be used (Johnson-Delaney, 2002).

Injectable anaesthetics have been used in prairie dogs. However, care should be taken, particularly in obese animals that may have variable responses to injectable agents.

Supportive care

Supplemental heating is necessary during anaesthesia to prevent hypothermia in chipmunks and prairie dogs. The patient's rectal temperature should also be monitored. Similarly to other small species, fluids and nutritional support are often required during hospitalised sciuromorphs.

SUBORDER HYSTRICOGNATHI

Guinea pigs (*Cavia porcellus*), chinchillas (*Chinchilla laniger*) and degus (*Octodon degus*) are hystricomorph rodents. These species are monogastric herbivores. Most animals are sufficiently calm to allow a conscious physical examination, although individual animals in a debilitated condition may become too stressed to complete the examination at one time.

Family Cavidae

Guinea pigs are sociable and housing a companion with the patient may encourage normal behaviour. The use of bedding such as shredded newspaper or cardboard hide boxes will also reduce stress. Dietary provisions should include good-quality hay, a selection of vegetables including leafy greens, proprietary guinea pig concentrate pellets or mix, water in a bowl or bottle (depending on what the individual is accustomed to), with vitamin C supplementation (at 2 g/L of drinking water (Quesenberry, 1994) or 10–30 mg/kg/day orally (Adamcak and Otten, 2000)).

Temperature

This species conserves heat well, but is prone to heat stress. Ideally, the environmental temperature should be 18–26°C (Harkness and Wagner, 1995c). However, as with other small mammals, supplemental heat should be provided to guinea pigs during anaesthesia. Careful monitoring of core temperature with a rectal probe (Fig. 4.7) should be performed during anaesthesia and during the recovery period. Guinea pigs are prone to heat stress and care should be taken to avoid overheating animals during hospitalisation.

Cardiovascular system

Guinea pigs have 70–75 ml of blood per kilogram body weight. The short neck of the guinea pig comprises a thick layer of muscle ventrally, making jugular venepuncture difficult. A cut-down technique should be used if a catheter is to be placed in the jugular vein (Quesenberry et al., 2004). For administration of fluids, intravenous sites



Figure 4.7 • Measurement of rectal temperature in a guinea pig, *Cavia porcellus*, using a digital thermometer.

available in this species are the lateral saphenous vein or cephalic vein, although these small veins are difficult to catheterise. A 22–25-gauge needle or 24-gauge or smaller catheter should be used. The ear veins are visible, but very small, and hence difficult to access in guinea pigs. An alternative site for phlebotomy or administration of drugs is the anterior vena cava, for which sedation or general anaesthesia is required. If long-term intravenous access is required, venous access ports can be used. Routinely, fluids are administered subcutaneously (avoiding the interscapular region where the skin is closely apposed to underlying tissues, including the subcutaneous fat pad) or intraperitoneally (Brown and Rosenthal, 1997).

The large guinea pig heart lies midline at the level of the second to fourth intercostals space, with a narrow space bilaterally for lungs (Breazile and Brown, 1976). In immature animals the cranial mediastinum contains the cervical thymus, being replaced by fat in the adult (Harkness and Wagner, 1995a). These anatomical relationships mean that the lungs are very small in guinea pigs and, hence, any lung pathology may readily cause clinical signs or increase the risk of anaesthesia in this species.

Respiratory system

The opening into the oral cavity is narrow and, as with all rodents, the oral cavity is long. Passage from the oropharynx to the pharynx and thence into the respiratory tract is via the palatal ostium (or interpharyngeal ostium), which is the central opening between the caudal tongue and the soft palate (Timm et al., 1987). Endotracheal intubation is possible in the guinea pig, but difficult, as the palatal ostium is a small opening, visualisation is poor and lateral deviation when introducing the tube will damage the vascular velopharyngeal folds in the soft palate (Quesenberry et al., 2004). An otoscope can be used to visualise the glottis and insert a guide wire, and the otoscope removed before threading an endotracheal tube (16–12-gauge catheter) over the wire (Flecknell, 1996b).

Pneumonia is common in pet guinea pigs, with damp or humid environments predisposing to bacterial infections, such as *Bordetella bronchiseptica* and *Streptococcus pneumoniae*. Viral pneumonia has also been reported. Primary pulmonary neoplasia, bronchogenic pulmonary adenoma, is common in guinea pigs. Lymphosarcoma, caused by a type C retrovirus, may affect the mediastinal lymph nodes and cause dyspnoea (Collins, 1988). Supportive care should be administered before anaesthesia is induced in dypnoeic animals, including oxygen therapy, fluid administration and oral vitamin C (O'Rourke, 2004).

Digestive system

Guinea pigs should be fed hay and fresh vegetables, supplemented with a concentrate mix (preferably complete pelleted diet rather than a cereal mix). They have a daily requirement for vitamin C of approximately 10 mg/kg, rising to 30 mg/kg/day during pregnancy. Good-quality food should always be available to hospitalised guinea pigs. Unfortunately, many will become depressed and refuse to eat and drink while hospitalised, so assist feeding (Table 4.2) is often necessary. Proprietary herbivore formulas (for example, Oxbow® Critical Care for Herbivores, Petlife International Ltd, Bury St Edmunds, Suffolk), softened guinea pig concentrate pellets, or vegetable baby food (dairy-free) can be administered orally (Quesenberry et al., 2004). Vitamin C should be given daily to hospitalised animals, either in the drinking water or directly administered by syringe if the patient is not drinking. Housing a companion simultaneously may reduce stress and encourage normal behaviour in these social animals, but can make assessment of appetite and urine and faecal production difficult.

The gastric emptying time in guinea pigs is normally 2 h and total gastrointestinal transit time 20 h on average, longer if coprophagy is included (Jilge, 1980). As this species has a very small lesser stomach curvature, they cannot vomit, and fasting is not required before anaesthesia. Guinea pigs feed primarily at dawn and dusk, but often retain food in their oral cavity. For this reason, the oral cavity should be checked for the presence of food material and cleaned with cotton-tipped swabs on induction of anaesthesia if necessary.

> Check the oral cavity after induction and remove retained food if present to avoid aspiration.

Gastrointestinal hypomotility is common after anaesthesia or surgery in guinea pigs, or associated with other disease processes or stress. Prokinetics (see Table 2.3) are usually administered prophylactically when guinea pigs are anaesthetised to reduce the risk of ileus.

Diarrhoea in guinea pigs may be caused by a number of aetiologies, including bacterial overgrowth secondary to oral administration of certain antibiotics, primary bacterial enteritis and endoparasites (O'Rourke, 2004). The guinea pig should be stabilised before anaesthesia is induced, by correcting fluid deficits caused by diarrhoea and the common concomitant anorexia.

If hepatic disease is suspected, blood biochemistry may be performed. Alanine aminotransferase (ALT) is not sensitive or specific for hepatocellular damage in guinea pigs (White and Lang, 1989). Hepatic lipidosis is common after a period of anorexia, and may result in ketosis and hypercholesterolaemia (Quesenberry et al., 2004).

Urinary system

Daily water update is approximately 100 ml/kg in guinea pigs (Manning et al., 1984). The normal urine pH is 9.0 in these herbivores (Navia and Hunt, 1976). If an animal has been anorexic for a few days or more, dipstick analysis of urine can be used to assess for the presence of ketones. Ketonuria is produced in ketoacidotic animals, which will require stabilisation of metabolic derangements prior to anaesthesia.

Many guinea pigs older than 3 years of age have chronic interstitial nephritis, which may be associated with other

conditions, such as diabetes mellitus, or occur secondary to renal amyloidosis. Guinea pigs commonly develop urinary tract calculi. Post-renal azotaemia may result from partial or complete obstruction. Volatile agents, such as isoflurane or sevoflurane, are the anaesthetic of choice for investigation and surgical treatment of such cases (O'Rourke, 2004).

Endocrine system

Diabetes mellitus has been reported in some guinea pigs, some responding to insulin while others are non-insulin-dependent (Bowden, 1959; Hartmann, 1993; MacKay et al., 1949; Marlow, 1995). As is the case with other species, diabetes should be stabilised (with an appropriate diet and/or insulin) prior to anaesthesia. As guinea pigs are not fasted prior to anaesthesia, hypoglycaemia is less likely during the procedure, but blood glucose levels should be monitored throughout the anaesthetic and in the recovery period, and dextrose administered as required.

Reproductive system

Dystocia is common in guinea pigs, and Caesarean sections are often warranted. The anaesthetic of choice for this procedure is a volatile agent, either isoflurane or sevoflurane. Pre-medication with buprenorphine may be helpful in causing mild sedation before mask induction, and will also provide post-operative analgesia.

Another common reason for anaesthesia of guinea pigs is surgical excision of mammary neoplasia. A small number of these are malignant, for example adenocarcinomas. Metastasis is rare, but the thoracic cavity should be auscultated and radiographed to assess for pulmonary involvement and function. The abdomen should also be assessed for visceral involvement by palpation and ultrasound.

It is unwise to anaesthetise a guinea pig suffering from pregnancy toxaemia. The animal will be hypoglycaemic, ketonuric, proteinuric and aciduric (pH 5–6). Hepatic lipidosis also usually occurs. Despite intensive supportive care, many animals die (O'Rourke, 2004). Treatment is stressful, and restraint and anaesthesia will add to the stress and thereby speed mortality.

> Guinea pigs with pregnancy toxaemia are very poor candidates for anaesthesia.

Induction and maintenance of anaesthesia

Halothane may cause hypotension and hepatic damage. Isoflurane is safer; however, irritation of mucous membranes during induction may cause lacrimation and salivation in guinea pigs (Flecknell, 2002). Sevoflurane causes less irritation to the airways. Doses for injectable sedatives or anaesthetic protocols are shown in the tables (see Tables 4.4 and 4.6), but there is much individual variation in response to these drugs.

Anaesthesia is usually induced in guinea pigs with a volatile anaesthetic in an induction chamber. A period of preoxygenation precedes addition of the anaesthetic agent. Once the righting reflex is lost, the guinea pig is removed from the chamber and oxygenation (for short procedures) or anaesthesia (for longer procedures) maintained via a facemask. It is important to use a small mask to minimise dead space, and for the mask to be close-fitting to reduce contamination of the environment with waste gases.

Volatile anaesthetic agents are primarily used for short investigative procedures in guinea pigs. However, there are two scenarios where injectable agents are preferable. It may be difficult to maintain sufficient depth of anaesthesia for surgery using inhalation agents alone, or the procedure to be performed may require access to the head that is restricted by a facemask.

During anaesthesia, guinea pigs frequently become apnoeic. This can make maintenance of anaesthesia difficult via volatile agents solely (Flecknell, 2002). In this scenario, injectable sedatives (see Table 4.4) may be used to relax the patient so a normal respiratory pattern resumes, or injectable anaesthetics (see Table 4.6) administered to replace the requirement for inhalational agents. If injectable agents are used alone to provide anaesthesia, oxygen should always be supplemented via a facemask.

Two common reasons for anaesthetising guinea pigs are for dental treatment, necessitating access to the oral cavity, or the treatment of cervical lymphadenitis. In the former case, it may be possible to intubate the patient, but the endotracheal tube and attached anaesthetic circuit will make the dental procedure difficult. Similarly, when operating on the cervical region, a facemask may intrude on the sterile surgical field. It is easier to use injectable anaesthetic agents and to provide supplemental oxygen via a small mask over the nose (Fig. 4.8). If necessary, anaesthetic gases can be administered via the nares, but a good seal between mask and patient may not be achievable, allowing environmental contamination.



Figure 4.8 • Anaesthetised chinchilla, *Chinchilla laniger*, maintained with isoflurane via a closely fitting facemask.

Epidural anaesthesia has been reported in the guinea pig (Thomasson et al., 1974).

Anaesthetic monitoring

The heart rate can be palpated or auscultated using a bell stethoscope over the thoracic wall, but rates up to 300 beats per minute are extremely difficult to count. In larger patients, an oesophageal stethoscope may be used similarly. Echocardiogram (ECG) pads can be attached to the feet (Fig. 4.9) or needle probes placed subcutaneously to inhance electrical conduction (Schoemaker and Zandvliet, 2005), but many machines cannot detect the low signal strengths and high frequencies in these animals (Flecknell, 2002).

Respiration is usually monitored by observing the patient. If a close-fitting facemask is used, breathing movements may be seen in the reservoir bag. Respiratory monitors can be used, but care should be taken in the choice of equipment so as not to increase dead space within the anaesthetic circuit (see Chapter 1). Pulse oximeters may be attached to the paw, but the high heart rate in guinea pigs may again be greater than the limit on some models (Flecknell, 2002). Oxygen saturation is improved by administering oxygen via a facemask throughout anaesthesia.

As guinea pigs are not routinely intubated during anaesthesia, PPV is not usually possible. It can be attempted using a tightly fitting facemask by compressing the reservoir bag with the expiratory valve temporarily closed; however, inadvertent oesophageal insufflation may result in gastric tympany. Alternative methods of respiratory assistance are thoracic compression and the use of respiratory stimulants, such as doxapram (Flecknell, 2002).

Monitoring and maintenance of body temperature are essential in guinea pig anaesthesia. Supplemental heat can be provided as for other species, and a rectal thermometer (see Fig. 4.7) used to monitor temperature. These processes should be continued during the post-anaesthetic period, until the guinea pig has recovered sufficiently to be able to thermoregulate.



Figure 4.9 • Echocardiograph pads on an anaesthetised guinea pig, *Cavia porcellus*. The pads are stabilised on the small feet using adhesive tape.

Supportive care

Since intravenous access is limited in the guinea pig, fluids to support the circulation are usually administered as a bolus subcutaneously (see Fig. 4.1) or intraperitoneally (see Fig. 4.2) during anaesthesia. Subcutaneous fluids are more slowly absorbed.

During recovery, a facemask can be used initially, moving to a chamber supplemented with oxygen if necessary when the animal becomes more reactive. In the recovery period, supplemental heat should be continued until the patient is able to thermoregulate. If volatile agents have been used, recovery is usually rapid. Injectable agents produce a more prolonged recovery, as will painful procedures. If recovery is unexpectedly slow, body temperature should be checked using a rectal thermometer and analgesia requirements reassessed.

It is important that these herbivorous animals begin eating soon after anaesthesia, to reduce the risk of ileus. Analgesia (Table 4.7) may be required if a painful condition exists or surgery has been performed. Opioids, non-steroidal anti-inflammatory drugs (NSAIDs), and local anaesthesia can all be used in guinea pigs (Flecknell, 2002). Prokinetics may be necessary to stimulate gastrointestinal motility, but often syringe feeding is more beneficial in maintaining hydration and movement of ingesta through the digestive tract.

Family Chinchillidae

Chinchillas usually occur in large groups in the wild. The more common situation in captivity is a single animal, a pair, or a polygamous group of a single male with two to six females (Quesenberry et al., 2004). As shy animals, provision of a cardboard box (as for guinea pigs) or plastic pipe hide will reduce the stress of hospitalisation. Pets should have climbing and jumping space at home, but a single-level kennel is satisfactory for hospitalisation purposes. A dust bath (using commercial chinchilla sand or volcanic ash) should be provided for a short time daily during hospitalisation.

Chinchillas are adept at hiding signs of disease and subclinical pathology is often present. It is, therefore, important to question the owner closely regarding husbandry conditions, to identify any factors that may predispose to illness. A full clinical examination is possible on most pet chinchillas, and disease processes that the owner has not noticed may be detected in this manner.

Temperature

Chinchillas are adapted to living in the cold temperatures of the Andes mountains and have thick fur. The environmental temperature range should ideally be 10–20°C, although chinchillas are adapted to ambient temperatures of between 18.3°C and 26.7°C, with relative humidity below 50% (Donnelly, 2004a; Webb, 1991). Chinchillas do not tolerate damp or wet environments (Quesenberry et al., 2004). Although hypothermia is the main concern during

anaesthesia, they may easily succumb to hyperthermia when environmental temperatures are above 28°C (Hoefer and Crossley, 2002). Environmental and rectal temperature must be monitored closely during anaesthesia and the recovery period.

Cardiovascular system

As with guinea pigs, intravenous access can be difficult and sedation or anaesthesia is required in most animals. 25-gauge needles or insulin syringes (28-gauge) can be used to access peripheral veins, such as the lateral saphenous or cephalic. Catheters should be 24-gauge or smaller. A cut-down technique is required for jugular access. If long-term intravenous access is required, venous access ports can be used (Quesenberry et al., 2004).

Cardiomyopathy and valvular disease have been reported in chinchillas (Hoefer and Crossley, 2002). If clinical signs are present, they are usually of dyspnoea associated with cardiac failure. Cardiac murmurs heard on auscultation may or may not be significant (Hoefer and Crossley, 2002). Echocardiography and electrocardiography are indicated to investigate any heart murmurs identified on clinical examination before the animal is anaesthetised (Donnelly, 2004a).

Gastrointestinal system

Chinchillas are hindgut fermenting herbivores. In the wild they consume a variety of grasses, cactus fruit, leaves and bark of small shrubs and bushes. The vegetation is tough and fibrous, with low energy content. The captive chinchilla diet should predominantly be good-quality meadow grass hay (for example, Timothy grass hay), with a small amount of proprietary chinchilla pellets, and *ad libitum* water. Occasional treats may include fruit and small amounts of greens (Hoefer and Crossley, 2002). Chinchillas eat mainly at night, so food should be available constantly. A daily weight check will be a more accurate method of assessing appetite than observations during daylight hours. The mean gastrointestinal transit time is 12–15 h (Quesenberry et al., 2004).

Dental disease is the most common reason for presentation of pet chinchillas at veterinary practices. Often animals have had a reduced or altered appetite for some time, and many animals are in poor body condition. In these cases, the chinchilla must be assessed and a decision made as to whether the anaesthetic required for dental treatment should be postponed while nutritional support is given, or whether the animal is stable enough to be anaesthetised and receive dental attention, which will relieve oral discomfort and allow the animal to self-feed. In some cases, a staged procedure is used, whereby the use of volatile agents or a short-acting combination allows an initial assessment and perhaps minor dental treatment. After a few days of nutritional support, when the chinchilla is in better body condition, a more prolonged procedure can be performed under a longer anaesthetic.

Diarrhoea may be caused by an inappropriate diet, overfeeding, sudden dietary change, bacterial or parasitic enteritis (Donnelly, 2004a). Hepatic disease caused by metronidazole toxicity and neoplasia (Nobel and Neumann, 1963) have been reported.

As with other herbivores, ileus can cause significant morbidity (and mortality in some instances) post anaesthesia. Prokinetics are frequently used in chinchillas perianaesthetically (see Table 2.3). Syringe feeding is also a useful procedure if the patient is not self-feeding soon after anaesthesia.

Urinary system

Normal chinchilla urine has a pH of 8.5, and it is usually concentrated with a specific gravity greater than 1.045 (Merry, 1990). As is the case with guinea pigs, dipsticks can be used to check for ketonuria.

Calcium oxalate crystals may precipitate in the renal tubules, causing renal dysfunction (Goudas and Lusis, 1970). Lower urinary tract disorders, such as calculi, may lead to post-renal azotaemia. In animals with suspected urinary tract disease, renal function should be assessed before anaesthesia by analysing urine and blood parameters. If renal dysfunction is found, fluids should be administered before, during, and after anaesthesia to ensure renal circulation is not compromised. Drugs that may be metabolised or excreted via the kidneys should be avoided. Volatile agents, such as isoflurane and sevoflurane, may be used, as their excretion is almost completely via the respiratory tract.

Endocrine system

Diabetes mellitus has been reported in a chinchilla (Marlow, 1995). Glucosuria and ketonuria were present in the case, along with hyperglycaemia. Blood glucose levels should be monitored in diabetic animals perianaesthetically, encouraging them to feed normally as soon as possible when recovered.

Nervous system

Pre-anaesthetic clinical examination of chinchillas should include assessment of their demeanour and neurological function. Differential diagnoses for animals with clinical signs consistent with central nervous system disease include infection with viruses (for example, herpesvirus (Goudas and Giltoy, 1970; Wohlsein et al., 2002)), bacteria (for example, *Listeria monocytogenes* (MacKay et al., 1949)), protozoa (for example, *Frenkelia microti* (Dubey et al., 2000)), or nematodes (for example, *Baylisascaris procyonis* in Canada (Sanford, 1991)). Head trauma could also cause central nervous dysfunction.

Anaesthesia may adversely affect animals with a compromised central nervous system, primarily by reducing blood oxygen saturation and its supply to the brain. Care should be taken with these cases to provide sufficient oxygen, and to maintain the circulation and blood pressure

by administering fluids. Volatile anaesthetic agents are used in these cases, as they have the least depressive effects on metabolism and depth of anaesthesia can rapidly be altered if necessary. Midazolam or diazepam can be administered as a pre-medicant to reduce the risk of seizures.

Sedation and anaesthesia

Chinchilla sedation may be necessary for phlebotomy or non-painful procedures, such as radiography or ultrasonography. Midazolam can be used to produce mild sedation, or ketamine added to produce deeper sedation. The midazolam and ketamine mix can be used for pre-medication or light anaesthesia prior to induction/maintenance using gaseous anaesthetic agents (see Tables 4.4 and 4.6).

Induction of anaesthesia in chinchillas is commonly performed in a chamber using inhalational agents, often without pre-medication. After preoxygenation for a few minutes, 2–5% isoflurane or halothane is added to induce anaesthesia. Loss of the righting reflex denotes anaesthesia. Usually 2–4% isoflurane or 2–3% halothane is required for maintenance of anaesthesia (Hoefer and Crossley, 2002).

In some cases, injectable anaesthesia is preferable to gaseous agents, for example when dental disease necessitates access to the oral cavity. General anaesthesia can be induced using a mix of acepromazine and ketamine. This rapidly results in surgical anaesthesia that lasts for up to 1 h. Ketamine has a wide safety margin. Acepromazine should be avoided in hypovolaemic animals, and the doses listed for both drugs may be reduced for debilitated animals. The combination is not reversible, and sleep time can be up to 5 h (Morgan et al., 1981). Ketamine can also be used in combination with xylazine or diazepam. The ketamine combinations can be topped up with volatile agents, such as isoflurane, if necessary.

A study comparing three injectable anaesthetic combinations (Henke et al., 2004) showed the combination of midazolam with medetomidine and fentanyl to produce safer anaesthesia. Recovery after xylazine with ketamine anaesthesia was more prolonged. Cardio-respiratory depression was less compared to animals given ketamine with xylazine or medetomidine, and the triple combination protocol allowed complete and rapid reversal using antagonists. Bradycardia associated with alpha-2-agonists appears to be less marked in chinchillas than that seen in other species (Henke et al., 2004). This shorter recovery phase enables animals to return to normal physiological activity sooner, and reduces the risks of hypothermia and hypoglycaemia post anaesthesia.

Oxygen should be provided during all anaesthetics, usually via a small facemask (see Fig. 4.8). Where oral access is required, the end of the anaesthetic circuit may be held adjacent to the nares (Fig. 4.10) or a small nasal catheter used to administer oxygen.

Monitoring and supportive care

Chinchilla anaesthesia is monitored as for other small mammal species. The toe pinch withdrawal is the most reliable tool for monitoring depth of anaesthesia. The eyes should be coated with ocular lubricant (for example, liquid paraffin) to protect them from trauma, particularly when ketamine combinations are used, which result in open eyelids. During recovery, a soft surface, such as a towel, should cover food or bedding material that may be irritant to the eyes.



Figure 4.10 • Rat, *Rattus norvegicus*, with end of T-piece circuit used as a facemask to allow access to the submandibular region for surgery.

Supplemental heat should be provided during anaesthesia and in the recovery period until the patient is able to thermoregulate. It is useful to monitor body temperature using a well-lubricated rectal thermometer until the patient is mobile enough to move away from a heat source.

Unless a very brief gaseous anaesthesia has been performed, fluids are administered at the time of anaesthesia to support the circulation. Warmed fluids are usually administered subcutaneously; the intraperitoneal route can also be used.

If the chinchilla may be in discomfort, analgesia should be administered. Pain is likely to cause anorexia and result in ileus. Nutritional support is provided with prokinetics (see Table 2.3) and syringe feeds (see Table 4.2) as for other small mammals. As soon as the chinchilla has recovered sufficiently, good-quality hay is provided to encourage a return to normal appetite.

Family Octodontidae

Most techniques used in guinea pigs and chinchillas are appropriate for degus (see Fig. 4.3), such as venepuncture, as are doses for drugs and other treatments (Johnson-Delaney, 2002).

Respiratory system

Pneumonia is commonly seen in pet degus (Donnelly, 2004b). Primary respiratory tract neoplasia has also been reported (Anderson et al., 1990).

Digestive system

The degu is a herbivorous hind-gut fermentor. In the wild, they eat grass, leaves, bark, herbs, seeds, fruits, fresh cattle or horse droppings, and crops. Captive animals eat rodent chow, grass hay and occasional fresh greens. Inappropriate diets may lead to obesity (Donnelly, 2004b) or predispose to dental disease. Hepatocellular carninomas have been reported (Montali, 1980; Murphy et al., 1980).

Urinary system

Degus do not require much water, but should have water available *ad libitum* (Donnelly, 2004b).

Endocrine system

Amyloidosis of Langerhans islets may lead to diabetes mellitus in degus. This may be associated with certain viral infections or hyperglycaemia due to an inappropriate diet (Fox and Murphy, 1979; Najecki and Tate, 1999; Spear et al., 1984). Blood glucose levels should be closely monitored in these animals before and during anaesthesia, and in the recovery period. Intravenous dextrose can be administered if required.

Anaesthesia

The easiest option for anaesthesia of degus is complete inhalational anaesthesia (see Table 4.5). This is relatively safe and good for short, minor procedures, such as oral examination, phlebotomy and radiography. It is less useful for dental treatment or surgery on the head (which may interfere with facemask positioning).

For these latter procedures, injectable agents should be used as in the other hystricomorphs (see Tables 4.4 and 4.6). Options include sedation with midazolam, and anaesthesia with ketamine combinations (acepromazine, diazepam, or medetomidine). The degu should be accurately weighed on digital scales (see Fig. 1.9) to reduce the risk of overdose.

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5 Ferret anaesthesia

INTRODUCTION

The ferret, *Mustela putorius furo*, is commonly kept as a pet or working animal. Sedation or anaesthesia may be required to perform investigative procedures or surgery.

ANATOMY AND PHYSIOLOGY

Temperature

The normal ferret body temperature is 37.8–40°C (Fox, 1998). Ferrets do not have sweat glands, and they are vulnerable to heat stress above 32°C, particularly if humidity is also high (Brown, 2004; Lewington, 2005). The environmental temperature should not exceed 21.2°C for nesting jills (Bell, 2004).

Cardiovascular system

The heart lies obliquely between the sixth and eighth ribs, with the apex beat to the left. This caudal positioning of the heart makes cranial vena cava puncture a safer technique in ferrets compared to other species (An and Evans, 1998). In the healthy animal, the heart should not normally rest on the sternum (Brown and Rosenthal, 1997). The normal resting heart rate is 180–250 beats per minute (Petrie and Morrisey, 2004).

Mean systolic arterial blood pressure is 133 mmHg in the conscious jill or 161 mmHg in the hob. In the anaesthetised ferret, the mean diastolic arterial blood pressure is 110–125 mmHg (Fox, 1998). A sinus arrhythmia may be found in normal ferrets (Quesenberry and Orcutt, 2004), as may second-degree atrioventricular (AV) block (Petrie and Morrisey, 2004). Capillary refill time should be less than 2 s, and mucous membranes should be pink. Peripheral pulses are not easily palpable in ferrets, but an ultrasonic Doppler flow detector may be used to assess blood pressure indirectly or urine output may be used to assess cardiac output (Lucas, 2000).

Cardiac disease is prevalent in ferrets, who are susceptible to both dilated and hypertropic cardiomyopathy, and *Dirofilaria immitis* (Lewington, 2005). If possible, animals with cardiac disease should be given medications to improve cardiac function prior to anaesthesia, for example using furosemide, digoxin, and/or enalapril as appropriate (Schoemaker, 2002).

Total blood volume is usually 5–7% of body weight, and is approximately 40 ml in a jill and 60 ml in a hob (Fox, 1998). Common venepuncture sites in the ferret are the cephalic, lateral saphenous and jugular veins. The jugular vein lies quite laterally on the neck (O'Malley, 2005). The ventral coccygeal artery can also be accessed (Curl and Curl, 1985), as can the cranial vena cava in the anaesthetised animal (Schoemaker, 2002).

Ferrets have a relatively high haematocrit compared to other species, at 46–61% (Petrie and Morrisey, 2004). Anaemia may be caused by blood loss, or a number of chronic diseases may lead to anaemia, and the risk of anaesthesia to the patient will depend on the aetiology. Animals with a packed cell volume (PCV) of less than 25% are likely to benefit from a blood transfusion.

Respiratory system

As the ferret's tongue is mobile as in cats, it is easily pulled rostrally to allow visualisation of the glottis for intubation. The ventral space in the nasal conchae is very narrow allowing passage of a catheter with a maximum diameter of 3.0 or 3.5 French if necessary for oxygen supplementation (Lewington, 2005).

Compared to other mammals, the ferret's thoracic cavity is large. The long lungs have a correspondingly large total lung capacity (Lewington, 2005). Diaphragmatic movement is more important in ventilation of the anaesthetised ferret than costal movement.

Ferrets may sneeze during clinical evaluation. This is often in response to dust or debris inhalation, and should not

be of concern unless it becomes frequent or other clinical signs are noted (Brown, 2004). Primary respiratory tract disease is relatively rare and includes viral (canine distemper virus, human influenza virus), rarely bacterial (for example, *Streptococcus zooepidemicus*, *S. pneumoniae*), parasitic (*Pneumocystis carinii*) and rarely mycotic (for example, *Blastomyces dermatitidis*, *Coccidioides immitis*) infections. Aleutian disease virus may cause an interstitial pneumonia in young animals. Differential aetiologies for a dyspnoeic ferret include cardiac disease (see above), thoracic trauma, neoplasia (usually metastases), gastrointestinal disease, such as megaoesophagus (with laboured breathing due to aspiration pneumonia) or gastric bloat (Hoefer and Bell, 2004).

Pleural effusion may be present in animals with cardiomyopathy or lymphoma, further compromising the patient during anaesthesia (Schoemaker, 2002). In these cases sedation or anaesthesia may be required to investigate the disease process.

BOX 5.1 Cardiovascular and respiratory systems

- Relatively large thoracic cavity
- Heart quite caudal in thorax
- Normal heart rate 180–250 beats per minute
- Normal blood volume 5–7% body weight
- Cardiac disease common; primary respiratory tract disease rare

URINARY SYSTEM

Normal water intake is 75–100 ml (Moody et al., 1985), producing 26–28 ml of urine daily (Fox, 1998). Normal urine pH is 6.0–7.5 (Quesenberry, 1996; Thornton et al., 1979). In some patients, blood biochemistry parameters may be assessed prior to anaesthesia. Blood urea nitrogen (BUN) levels are affected by renal and non-renal factors, and do not elevate simultaneously with serum creatinine in renal failure (Hillyer, 1997). Neither BUN nor serum creatinine levels increase until the kidney is 75% damaged, and so are relatively insensitive assessments of renal function, but small elevations are often significant (Esteves et al., 1994).

Renal disease may not cause clinical signs in ferrets. However, significant numbers of animals will have some degree of renal dysfunction, for example chronic interstitial nephritis in older animals (Kawasaki, 1994). Urolithiasis may lead to post-renal azotaemia. Where they are present, clinical signs of urinary tract disease are similar to those seen in other species (Pollock, 2004).

Azotaemic animals are usually anaesthetised with isoflurane. An alternative is to use ketamine with xylazine, reversing the xylazine with yohimbine or atipamezole. For this protocol it is advisable to administer fluids intravenously or subcutaneously before anaesthesia (Bell, 2004).

Gastrointestinal system

Ferrets are carnivorous. Their diet is low in carbohydrate and fibre, containing 9–28% fat. In the wild they consume whole carcases. Most captive animals are fed formulated diets containing 30–35% animal protein, in addition to chicks, mice, rats and raw egg (Brown, 2004). Water is provided in a bottle or weighted bowl. Ferrets eat 140–190 g of food daily (Fox, 1998), and have a rapid gastrointestinal transit time of 3–4 h in the adult animal (Bell, 1999). Unlike most of the other species discussed in this section, ferrets are able to vomit and are, therefore, fasted prior to anaesthesia.

Diarrhoea in ferrets may be due to various causes, from dietary indiscretion and infectious agents to inflammatory bowel disease and severe metabolic disorders (Hoefer and Bell, 2004).

Hepatic disease is common in ferrets, which may affect anaesthetic drug metabolism. Neoplasia is the predominating aetiology, in particular lymphoma. Chronic anorexia may lead to hepatic lipidosis, as may chronic gastrointestinal disease. Elevated alanine aminotransferase (ALT) is usually found on biochemistry in ferrets with hepatic disease, sometimes with elevated alkaline phosphatase (ALP) (Hoefer and Bell, 2004).

Endocrine system

Many disease processes can affect the ferret endocrine system, which may affect the patient's physiological responses to anaesthesia.

Adrenal gland pathology usually causes secretion of sexual hormones from the cortical region, leading to lethargy and muscle atrophy among other clinical signs (Lewington, 2005). Periurethral cysts may occur in male ferrets with adrenal gland disease. These cysts may obstruct urinary outflow, leading to metabolic abnormalities requiring stabilisation prior to adrenal gland surgery. Catheterisation of the bladder may be difficult without drainage of the cysts. Anaemia and pancytopenia may also occur (similar to oestrogen toxicosis). Many animals with adrenocortical disease have concomitant insulinomas and splenomegaly. As these are usually older animals, remember to check for cardiac disease or lymphoma in these cases. Cardiac disease is a common cause of peri-operative mortality in adrenalectomy surgeries (Lawrence et al., 1993; Rosenthal et al., 1993; Weiss and Scott, 1997; Weiss et al., 1999).

Adrenal medulla disease may occur in the form of phaeochromocytomas. These produce excess catecholamines, and affect the cardiovascular system. Clinical signs include tachycardia, dyspnoea, and cardiovascular collapse (Quesenberry and Rosenthal, 2004).

Oestrogen toxicosis may occur in females either with persistent oestrous (Sherrill and Gorham, 1985) or adrenal disease (de Wit et al., 2001). Haematopoietic tissue is affected, with a predominant finding of non-regenerative anaemia and leukopenia (Purcell and Brown, 1999; Rosenthal, 1994).

Insulinomas are relatively common in pet ferrets, and resulting hypoglycaemic crises should be stabilised before

anaesthesia is instigated. Other disease processes that may cause hypoglycaemia in ferrets are starvation, sepsis and hypoadrenocorticism (Ludwig and Aiken, 2004). Clinical signs include hindlimb paresis and central nervous system signs that may result from associated brain dysfunction. Hypoglycaemia may produce sinus bradycardia.

Diabetes mellitus has been reported in ferrets, most commonly after pancreatic surgery for removal of insulinomas (Quesenberry and Rosenthal, 2004).

BOX 5.2 Common endocrine diseases in ferrets that may cause metabolic or haematological changes affecting anaesthesia

- Adrenal gland disease
- Insulinoma
- Persistent oestrus
- Diabetes mellitus

Nervous system

If central nervous system abnormalities are found in the clinical examination, anaesthesia should preferably be postponed until the aetiology has been identified. Many pathologies will affect the patient's response to and risk from anaesthesia. If possible, stabilise the patient prior to anaesthesia. Causes of paresis or seizures in ferrets include hypoglycaemia associated with insulinomas, cardiac disease, metabolic derangements, toxins (for example ibuprofen), gastrointestinal disease, primary neurologic disease (for example neoplasia, intervertebral disc disease), Aleutian disease, rabies or late-stage canine distemper virus infection. Central nervous system disease includes trauma, infection, inflammation or neoplasia (Antinoff, 2004).

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

A history should be taken and a full clinical examination of the conscious animal undertaken to identify the extent of any disease processes before sedation or anaesthesia. Findings will help ascertain whether the animal is likely to survive anaesthesia and allow appropriate selection of anaesthetic agents.

Hospitalisation facilities

As their ancestors' natural behaviour was to live in underground burrows, ferrets prefer to sleep in an enclosed area and have digging opportunities when hospitalised (Brown, 2004). This can be provided with soft bedding, such as towels and shredded paper. Care should be taken to ensure that cage bars are sufficiently close to prevent escapes (Quesenberry and Orcutt, 2004).

Proprietary ferret foods are available, but cat foods are similar and can be fed to ferrets for short periods during hospitalisation.

Fluid and nutritional support

Hydration should be maintained during hospitalisation, including replacement of existing deficits and ongoing losses. Fluid therapy is usually administered subcutaneously or intraperitoneally (Table 5.1). Intravenous or intraosseous access is preferable for ill animals (Quesenberry and Orcutt, 2004). For intravenous catheterisation, the lateral saphenous and cephalic vein are most commonly used (Schoemaker, 2002).

Fasting

Ferrets should be fasted for 4 h prior to planned procedures to reduce the risk of vomition or regurgitation and aspiration (Schoemaker, 2002).

EQUIPMENT REQUIRED

Endotracheal tubes ranging in size from 2 mm to 4 mm may be used in ferrets, depending on the size of the animal. A laryngoscope is useful for intubation.

TECHNIQUES

Routes of administration

Fluids and drugs are given to ferrets similarly to other small mammals. Table 5.2 lists injection sites for ferrets.

Intubation

Endotracheal intubation in ferrets is similar to the procedure in cats. Local anaesthetic is sprayed on to the glottis and time allowed for anaesthesia to occur, before passage of an uncuffed endotracheal tube (diameter 2–4 mm for adult ferrets).

PRE-ANAESTHETICS

Medetomidine can be used to cause light sedation, either for minor procedures or prior to induction with another agent. Atipamezole can be administered to reverse the medetomidine and speed recovery (Schoemaker, 2002).

Acepromazine can be used to produce sedation in ferrets, administered at 0.1 mg/kg subcutaneously or intramuscularly (Heard, 1993).

Table 5.1: Fluid and nutritional support for ferrets

FLUID	ROUTE	DOSE	FREQUENCY	INDICATION/COMMENT
Crystalloids, for example lactated Ringer's solution	IV, SC	60–70 ml/kg/day[3]	CRI (IV) or divide into 2–3 boluses (slow IV, SC)	Maintenance requirements (increase if fluid losses present) Support intravascular volume
Colloids, for example hydroxyethyl starch (hetastarch)[3]	IV	5 ml/kg	Bolus over 15 min, can repeat with total dose <20 ml/kg/day	Shock therapy
		10–20 ml/kg/day	As CRI	Improves intravascular fluid volume and oncotic pressure Can coadminister with crystalloids, reducing crystalloid volume at 33–50%
Blood transfusion[1]	IV, IO	6–12 ml/animal (collected from recipient at ratio of 6 ml blood into 1 ml anticoagulant, such as acid-citrate-dextrose)	–	Treatment of anaemia with variety of aetiologies Indicated if PCV <25% and clinical signs or requires surgery, or if thrombocytopenic with clinical signs No need to cross-match donor blood with recipient's
Haemoglobin solutions, for example Oxyglobin®, Biopure Corp., Cambridge, MA)	IV	6–15 ml/kg[2]	Infusion over a 4-h period, once or twice in a 24-h period	Anaemic animals
Liquidised diet: proprietary nutritional support diets (canine a/d for carnivores, Hill's®), baby food	PO	5–10 ml/animal	q8h	Anorexic animals Warm food first Use organic, lactose-free baby foods

Key: CRI = continuous rate infusion, IO = intraosseous, IV = intravenous, PCV = packed cell volume, PO = orally, q8h = every 8 hours
[1] (Hoefer, 1992); [2] (Orcutt, 2001); [3] (Quesenberry and Orcutt, 2004)

Table 5.2: Routes of drug administration in ferrets

SITE	TECHNIQUE	COMMENTS
Intramuscular	Quadriceps muscles, lumbar muscles	Very small muscle mass, so SC injections preferable
Intraosseous	Proximal femur, proximal tibia	Useful access to circulation in collapsed animals
Intraperitoneal	Caudal right abdominal quadrant	Collapsed or anaesthetised animals only Useful for fluid therapy

SITE	TECHNIQUE	COMMENTS
Intravenous:		CRI or divide into 2–3 boluses over day, maintenance = 60–70 ml/kg/day
Lateral saphenous vein Cephalic vein Jugular vein	Thick skin, so use cut-down technique Short 22–26 gauge over-the-needle catheter Light dressing to reduce risk of self-removal	First three are good sites for catheter placement; place when anaethetised; can be difficult to place and maintain catheter
Cranial vena cava	25 gauge 25 mm needle Dorsal recumbency, needle at 30–45° angle into thoracic inlet between manubrium and first rib, direct needle to opposite hindleg	Anaesthesia usually required
Ventral coccygeal artery	21–20-gauge needle Insert needle at 30–45° angle towards body, into ventral tail groove midline, to depth of 2–3 mm	Topical anaesthesia (lidocaine [lignocaine]/ prilocaine)
Vascular access ports	Indwelling intravenous catheter with SC injection port	Surgical implantation required
Subcutaneous	Scruff	–

Key: CRI = continuous rate infusion, SC = subcutaneous (Quesenberry and Orcutt, 2004; Schoemaker, 2002)

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Injectable agents

After medetomidine sedation, intravenous *propofol* can be used to induce anaesthesia, prior to maintenance with gaseous agents. An alternative protocol is intramuscular *medetomidine and ketamine* (Table 5.3).

Volatile agents

Isoflurane is commonly used to induce anaesthesia for short procedures or in debilitated animals. Anaesthesia is induced using a facemask or an induction chamber with 4% isoflurane and maintained with 2% isoflurane (Schoemaker, 2002). Isoflurane decreases haematological parameters (maximally 15 min after induction) (Marini et al., 1994). For prolonged procedures, the ferret should be intubated. Local anaesthetic is applied to the larynx to reduce the risk of laryngospasm, before insertion of a 2–4 mm uncuffed endotracheal tube.

Isoflurane anaesthesia results in splenic sequestration of red blood cells. This may significantly reduce the haematocrit and plasma protein levels (Marini et al., 1997), but these return to baseline values less than 1 h after anaesthesia (Ludwig and Aiken, 2004). If required to investigate respiratory or gastrointestinal disease, for example to obtain radiographs or in azotaemic patients, isoflurane anaesthesia produces fewer side effects than other agents.

Anaesthetic maintenance

Most ferrets can be intubated. This allows oxygen supplementation to patients that have received injectable anaesthetic agents. It also allows controlled provision of volatile agents, including positive pressure ventilation (PPV) if required. If an animal cannot be intubated or a very short procedure is planned, a close-fitting facemask may be used to provide oxygen and/or volatile agents (see Fig. 5.1).

Recovery

Some injectable agents may be reversed, for example medetomidine with atipamezole. Extubation should be performed when the patient is showing signs of recovery, for example swallowing. If the ferret has been anaesthetised for more than a brief period, supplemental heating should be provided until normal homeostatic thermoregulation returns.

Suggested anaesthetic protocols

For brief procedures, such as venepuncture or radiography, mask or chamber induction with isoflurane or sevoflurane will provide rapid induction and recovery from anaesthesia.

Table 5.3 Sedative and anaesthetic agents in ferrets

DRUG	DOSE (mg/kg)	ROUTE	COMMENT/INDICATION
Acepromazine	0.1–0.5[8,15]	SC, IM	Pre-medication, light sedation
Atipamezole	1[7]	IM, SC, IV	Reversal of medetomidine
Atropine	0.04–0.05[9]	SC, IM, IV	Pre-anaesthetic; bradycardia, hypersalivation. Reduces xylazine's cardiovascular depressant effects[1]
Fentanyl/droperidol (Innovar-Vet®, Schering Plough)	0.15 ml/kg[6]	IM	Deep sedation or light anaesthesia
Fentanyl/fluanisone (Hypnorm®, Janssen)	0.3 ml/kg[5]	IM	Anaesthesia
Glycopyrrolate	0.01[9]	IM	Pre-anaesthetic; bradycardia, hypersalivation
Halothane[8]	3–3.5% 0.5–2.5%	Inhal	Induction Maintenance
Isoflurane[2]	 5% 2–3%	Inhal	Preferred volatile agent. Induction Maintenance
Ketamine[8]	 10–20 30–60	IM	Higher doses may produce apnoea Tranquillisation Anaesthesia
Ketamine + acepromazine	20–35 + 0.20–0.35[10,15]	SC, IM	Anaesthesia
Ketamine + diazepam	10–20 + 1–2[10]	IM	Anaesthesia (poor analgesia) [12]
Ketamine + medetomidine	5.00 + 0.08[5] 8.0 + 0.1[7]	IM	Induction Anaesthesia; hypotension and respiratory depression may occur
Ketamine + medetomidine + butorphanol	5.00 + 0.08 + 0.10[5]	IM	Induction
Ketamine + midazolam	5–10 + 0.25–0.50[13]	IV	Induction
Ketamine + xylazine	10–25 + 1–2[10,12]	IM	May result in cardiac arrhythmias Reverse xylazine with yohimbine or atipamezole Start IV/SC fluids beforehand; avoid in ill animals
Medetomidine	0.08–0.20[3]	SC, IM	Light sedation; pre-medicant (0.1 mg/kg usually used)

DRUG	DOSE (mg/kg)	ROUTE	COMMENT/INDICATION
Medetomidine + butorphanol	0.08 + 0.10[11]	IM	Anaesthesia Monitor for respiratory and cardiovascular depression
Midazolam	0.3–1.0[4]	SC, IM	Mild sedation, pre-medication
Naloxone	0.01–0.03[3]	IM, IV	Opioid reversal
Propofol	1–3[16]	IV	Induction of anaesthesia, after sedation with medetomidine (<8 mg/kg unpre-medicated[3])
Tiletamine/zolazepam (Telazol®, Fort Dodge)	12–22[14]	IM	Light anaesthesia; prolonged recovery at higher doses, so rarely used
Xylazine	1[8]	SC, IM	Tranquillisation; cardio-pulmonary side effects common so rarely used
Yohimbine	0.5[3,17]	IV, IM	Reversal of xylazine

Key: IM = intramuscular, IV = intravenous, SC = subcutaneous

[1] (Bell, 2004); [2] (Brown, 1993); [3] (Cantwell, 2001); [4] (Carpenter, 2005); [5] (Evans and Springsteen, 1998); [6] (Flecknell, 1987); [7] (Flecknell, 1997); [8] (Fox, 1988); [9] (Heard, 1993); [10] (Hillyer and Brown, 2000); [11] (Marini and Fox, 1998); [12] (Moreland and Glaser, 1985); [13] (Morrisey and Carpenter, 2004); [14] (Payton and Pick, 1989); [15] (Ryland et al., 1983); [16] (Schoemaker, 2002); [17] (Sylvina et al., 1990)



Figure 5.1 • After induction with injectable agents, supplemental oxygen may be provided to ferrets via a facemask or endotracheal tube.

The addition of injectable agents may smooth the anaesthetic process for more prolonged or painful procedures. For example, the addition of analgesics, such as buprenorphine or butorphanol, will also produce mild sedation, reducing stress associated with induction using volatile agents.

Special anaesthetic cases

Dehydration can be severe with *gastrointestinal disease*. This should be corrected prior to anaesthesia if possible, preferably using intravenous or intraosseous fluid administration. If anaesthesia is required to investigate the disease, for example to obtain radiographs, isoflurane anaesthesia causes the least side effects.

Adrenocortical disease is often treated with surgical excision of the enlarged gland(s). Intravenous access is necessary to provide circulatory fluid support peri-operatively, along with dextrose if necessary for hypoglycaemic animals. Animals should be encouraged to self-feed as soon as possible on recovery. Electrolytes are monitored during recovery, particularly if bilateral adrenalectomies have been performed, which may lead to hypoadrenocorticism. Cardiovascular function is usually monitored via urine output post-operatively, with urea and creatinine levels used to evaluate renal function if a reduction in urine output or hyperkalaemia is detected (Ludwig and Aiken, 2004).

Surgical excision is the treatment of choice with phaeochromocytomas in the adrenal glands. Due to the high levels of circulating catecholamines, the cardiovascular system should be supported during anaesthesia, by administering intravenous fluids during anaesthesia and the recovery phase.

Surgery (ovariohysterectomy) is usually necessary to treat *oestrogen toxicosis* associated with persistent oestrus.

Aggressive supportive care should be given to stabilise the patient prior to anaesthesia, possibly including blood transfusion(s) to treat anaemia (see Table 5.1) (Pollock, 2004; Purcell and Brown, 1999).

Insulinomas often require surgical excision and careful preparation for anaesthesia is necessary. Pre-anaesthetic preparation for pancreatic surgery will involve controlling clinical signs by achieving and maintaining normoglycaemia. Prednisolone and/or diazoxide may be used to manage clinical signs medically, along with frequent feeding. If a hypoglycaemic episode is causing collapse or seizures, the ferret may require intravenous dextrose (0.25–2.0 ml bolus of 50% dextrose slowly to effect, followed by a continuous infusion of 5% dextrose if required) or, rarely, diazepam.

Fasting should be for a maximum of 6 h prior to anaesthesia, and blood glucose monitored before, during and after anaesthesia. Placement of an intravenous catheter allows dextrose to be administered with maintenance fluids for 1–2 h prior to anaesthesia (Quesenberry and Rosenthal, 2004). Maintenance of systemic blood pressure throughout the procedure will ensure pancreatic perfusion, reducing the risk of pancreatitis (Nelson and Salisbury, 1994).

As fasting for 12–24 h post surgery is advisable, continue intravenous fluids (with 2.5–5% dextrose if necessary) and monitor blood glucose levels until the patient is eating normally (Quesenberry and Rosenthal, 2004). Post-operative hypoglycaemia can usually be managed with feeding, prednisolone and diazoxide (Ludwig and Aiken, 2004).

If anaesthesia is required in *diabetic* animals, the pre-anaesthesia fasting period should be minimised. Blood glucose is closely monitored during and after anaesthesia, intravenous access is maintained until self-feeding, and food intake is encouraged as soon as possible on recovery.

ANAESTHESIA MONITORING

Observations on the patient

Cardio-respiratory system

The heart rate and rhythm may be assessed using a bell or oesophageal stethoscope. Observing the patient and anaesthetic reservoir bag, and auscultating the lungfields using a bell stethoscope, will allow monitoring of the respiratory rate, depth and rhythm.

Central nervous system

Anaesthetic depth is assessed similarly to other small carnivores, including corneal and palpebral reflexes, jaw tone and withdrawal reflexes.

Anaesthetic monitoring equipment

Electrocardiograms (ECGs) can also be used to monitor heart rate, rhythm and electrical conduction. ECG clips are placed as in the dog or cat, to the skin overlying the elbows and stifles or to pads attached on the footpads. As with other exotic pets, needle probes placed subcutaneously may be used to enhance electrical conduction. Measurements from ECGs in conscious ferrets have been reported (Schoemaker and Zandvliet, 2005).

Blood pressure measurement may be useful, for example in hypovolaemic patients. The indirect method is commonly employed, using a pneumatic cuff and ultrasonic Doppler flow detector. The cuff is placed proximal to the carpus or tarsus, or at the tail base. After a small area of overlying hair is shaved and ultrasound gel applied to improve contact, the Doppler probe is used to detect blood flow in the artery (for example, using the digital branch of the radial artery on the forelimb) (Lichtenberger, 2004).

A rectal or ear *thermometer* can be used to monitor body temperature during anaesthesia and the recovery period.

PERI-ANAESTHETIC SUPPORTIVE CARE

Hypothermia is common in ferrets, so *supplemental heating* is necessary during both anaesthesia and the recovery phase (Schoemaker, 2002). If surgery is being performed the use of waterproof drapes reduces the risk of excessive wetting of the patient with fluids used intra-operatively, which may result in evaporative cooling of the patient.

The patient should be encouraged to *feed* normally as soon after anaesthesia as possible. This is of particular importance in cases of insulinoma, when blood glucose should be closely monitored and intravenous glucose supplemented if hypoglycaemia is detected. If prolonged anaesthesia or recovery is predicted, or if a jill with pregnancy toxaemia is anaesthetised for Caesarean section, dextrose should be administered (Bell, 2004).

Syringe feeding is often helpful if the ferret is not self-feeding soon after anaesthesia, particularly nursing jills. It is helpful to offer a variety of foods to encourage self-feeding. Oesophagostomy tubes can be placed if longer-term support is necessary (Fisher, 2001). Motility stimulants, such as cisapride (0.5 mg/kg PO q8 h) and metoclopramide (0.2–1.0 mg/kg PO/SC q6–8 h), may also assist with gastrointestinal function (Schoemaker, 2002).

Analgesia

Analgesia should be administered as appropriate to aid recovery (Table 5.4). Ferrets are sensitive to paracetamol (acetaminophen) toxicity (Court, 2001). Non-steroidal anti-inflammatory drugs may alter renal circulation and should be used with caution. Those with preferential cyclooxygenase 2 (COX-2) inhibition may cause fewer side effects (Quesenberry and Orcutt, 2004).

EMERGENCY PROCEDURES AND DRUGS

If apnoea occurs, holding the legs and tilting the ferret in a cranio-caudal rocking motion will stimulate diaphragmatic breathing (Lewington, 2005).

Table 5.4: Analgesics in ferrets

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Buprenorphine	0.01–0.03[2]	SC, IM	8–12	Reduces doses of medetomine and ketamine necessary for anaesthesia
Butorphanol	0.1–0.5[1]	IM	4	Analgesia and sedation
Carprofen	1–5[3]	PO, SC	12–24	
Flunixin	0.5–2.0[2]	SC, IV	12–24	Provide fluid support
Meloxicam	0.1–0.2[3]	PO	Not known	

Key: IM = intramuscular, IV = intravenous, PO = orally, SC = subcutaneous
[1] (Cantwell, 2001); [2] (Heard, 1993); [3] (Schoemaker, 2002)

Table 5.5: Emergency drugs in ferrets

DRUG	DOSE (mg/kg)	ROUTE	INDICATION/COMMENT
Adrenaline (epinephrine)	0.02[7]	IV, IM, SC, IT	Cardiac arrest
Atropine	0.02–0.04[3]	SC, IM	Bradycardia
Dexamethasone	4–8[3]	IM, IV	Shock
Diazepam[6]	1 0.5–1.0 mg/kg/h	IM, IV CRI (IV)	Seizures Can repeat boluses
Doxapram	1–11[2,4]	IV	Respiratory stimulant
Furosemide	1–4[1]	SC, IM, IV, PO	Diuretic (duration 8–12 h)
Glycopyrrolate	0.01[5]	IM	Bradycardia, hypersalivation

Key: CRI = continuous rate infusion, IM = intramuscular, IT = intratracheal, IV = intravenous, PO = orally, SC = subcutaneous
[1] (Bartlett, 2002); [2] (Besh-Williford; 1987); [3] (Brown, 1999); [4] (Flecknell, 1987); [5] (Heard, 1993); [6] (Hillyer and Brown, 2000); [7] (Morrisey and Carpenter, 2004)

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Anaesthesia of other small mammals

6

AFRICAN PYGMY HEDGEHOG

INTRODUCTION

A species increasing in popularity as a pet is the African pygmy hedgehog or four-toed hedgehog (*Atelerix albiventris*). This insectivorous species is nocturnal, hiding in burrows during the day and emerging at night to search for invertebrates (Reeve, 1994). The hedgehog's spines make procedures difficult in conscious animals. Their response to handling is to curl into a ball and general anaesthesia is usually required to perform a clinical examination on these animals.

ANATOMY AND PHYSIOLOGY

Temperature

The optimal environmental temperature for this species is 24–29°C, with a range between 23°C and 32°C (Ivey and Carpenter, 2004; Johnson-Delaney, 2002). Supplemental heating is, therefore, usually required even in conscious animals, but is particularly important for debilitated individuals. Excessively low or high temperatures induce torpor (Larsen and Carpenter, 1999). Chronic hypothermia will result in immune suppression and metabolic derangements. Humidity should be low, less than 40%, in the environment.

Cardiovascular system

Cardiomyopathy and congestive heart failure have been reported in hedgehogs (Johnson-Delaney, 2002; Raymond and Garner, 2000). Both will compromise cardio-pulmonary function during anaesthesia.

Respiratory system

Respiration should be silent, but a stressed individual may 'hiss' in defence or aggression (Ivey and Carpenter, 2004).

Inappropriate husbandry, in particular sub-optimal environmental temperatures, may predispose hedgehogs to disease of the respiratory tract. Toxins and irritants, such as wet cedar bedding or smoke, may irritate the respiratory system. Various infectious aetiologies have been reported to cause pneumonia, including *Bordetella bronchiseptica*, *Pasteurella multocida*, and *Mycoplasma* spp. (Smith, 2000). Parasites and neoplasia (often metastatic spread to the lungs) may also cause respiratory disease (Hoefer, 1999; Raymond and Garner, 2001).

Urinary system

Renal diseases may affect a hedgehog's ability to metabolise and excrete anaesthetic drugs. Those seen in hedgehogs include tubular necrosis, infarcts, glomerulosclerosis, nephritis and glomerulonephropathies (Done, 1999; Raymond and White, 1999). A differential diagnosis for polydipsia and polyuria in a hedgehog would be Cushing's disease, although other clinical signs would be seen with this disease as in other species (Johnson-Delaney, 2002).

Digestive system

In the wild African pygmy hedgehogs are mainly insectivores and omnivores, with a high protein and low fat content, but they also consume some plants. In captivity, they are given cat or insectivore diets (proprietary 'hedgehog' diets also exist), mixed vegetables with calcium supplement and insects. Obesity is not infrequently seen in captive animals.

Enteritis is relatively common in hedgehogs, with infectious aetiologies including zoonotic *Salmonella* spp. (Craig

et al., 1997). If nutritional support is required, a protein-rich high-calorie canine or feline diet may be syringe-fed (Smith, 2000).

Hepatic lipidosis appears to be common in hedgehogs, particularly obese animals, with a variety of aetiologies (Raymond and White, 1999). Other disease processes that may affect hepatic function, and thereby drug metabolism, include neoplasia and human herpes simplex virus 1 (Allison et al., 2002; Lightfoot, 1999).

Nervous system, special senses

Several disease processes may affect the central nervous system, including torpor causing ataxia and systemic infections spreading to the nervous system (Ivey and Carpenter, 2004). Hedgehogs have acute senses of smell and hearing, including an ability to hear at ultrasonic frequencies.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

Many diseases result in non-specific clinical signs, which makes localisation and identification of pathological processes in hedgehogs challenging. Clinical examination is often unrewarding in the conscious patient. The history may alert the clinician to abnormalities, but anaesthesia is often required for clinical examination and further investigation.

Hospitalisation facilities

A quiet kennel area with subdued light will reduce stress in the hedgehog and aid recovery. Shredded paper should be provided to allow burrowing behaviour.

Fluid and nutritional support

Maintenance fluids are 50–100 ml/kg daily (Johnson-Delaney, 2002); these are usually warmed and injected subcutaneously.

TECHNIQUES

Routes of administration

The administration of fluids and medication is difficult due to the distinctive integument of the African pygmy hedgehog. The oral route is not possible in most animals due to the curl response, unless medication is taken in food. Subcutaneous injections in the flank or intramuscular injections are possible in conscious patients, but sedation or anaesthesia is usually required for other routes (Table 6.1).

PRE-ANAESTHETICS

Preoxygenation is advisable, particularly if respiratory disease is suspected from initial clinical evaluation of the patient. *Atropine* pre-medication may reduce hypersalivation in response to isoflurane (Lightfoot, 1999). Agents such as *diazepam* 0.5–2.0 mg/kg intramuscularly (Smith, 1992) may be used to produce mild sedation or to treat seizures.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Volatile agents

Volatile agents, such as *isoflurane*, are ideal for induction of anaesthesia, particularly for short procedures, such as clinical examination. Induction is easiest performed in a chamber, as the head is often not visible in the protective curled position.

Injectable agents

Injectable anaesthetics have been reported in hedgehogs, but recovery is often prolonged (Pye, 2001; Smith, 2000).

Table 6.1: Routes of drug administration in African pygmy hedgehogs

ROUTE	COMMENTS
Intramuscular	Orbicularis muscle of mantle, or quadriceps muscles
Intraosseous	Proximal femur Difficult
Intraperitoneal	Caudal right abdominal quadrant Useful for fluid administration; anaesthetised or collapsed animals only
Intravenous:	All under general anaesthesia
Femoral vein	–
Lateral saphenous vein	–
(Cephalic vein)	–
(Jugular vein)	Easier in thin animals
(Cranial vena cava)	Risk of cardiac puncture
Subcutaneous	Flank at junction of furred skin and mantle ('true' subcutaneous with 9 cm spinal needle to enter past the spines)

(Johnson-Delaney, 2002, Ivey and Carpenter, 2004)

Ketamine has been used in combination with diazepam, medetomidine or xylazine to induce anaesthesia (Table 6.2). Tiletamine and zolazepam have also been used, but rough recoveries are reported.

Anaesthetic maintenance

Provision of oxygen and maintenance with volatile agents during anaesthesia are via a tightly fitting facemask or endotracheal tube. Endotracheal tubes of 1.0–1.5 mm diameter, intravenous catheters (with the stylet removed) or feeding tubes may be used for intubation (Ivey and Carpenter, 2004). It is important to protect the airway in this way, particularly during procedures involving the oral cavity, such as dentals.

ANAESTHESIA MONITORING

During induction, the hedgehog will uncurl. Thereafter, monitoring of cardio–respiratory parameters and reflexes is similar to other small mammals.

PERI-ANAESTHETIC SUPPORTIVE CARE

Analgesia

Analgesics should be used where discomfort is suspected (Table 6.3).

Table 6.2: Sedative and anaesthetic drugs for hedgehogs

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Atipamezole	0.3–0.5[8]	IM	Medetomidine reversal
Atropine	0.01–0.04[5]	IM, SC	Pre-medicant
Diazepam	0.5–2.0[6]	IM	Mild sedation; seizure treatment
Ketamine + diazepam	5–20 + 0.5–2.0[3]	IM	Anaesthesia
Ketamine + medetomidine	5 + 0.1[8]	IM	Anaesthesia
Ketamine + medetomidine + fentanyl	2 + 0.2 + 0.1[1]	SC	Anaesthesia; good muscle relaxation
Halothane	To effect[4]	Inhal	Less preferable than isoflurane
Isoflurane	3–5%[7] 0.5–3.0%[7]	Inhal	Induction of anaesthesia Maintenance of anaesthesia
Medetomidine	0.2[2]	IM	Heavy sedation
Naloxone	0.16[1]	IM	Fentanyl reversal
Sevoflurane	To effect[5]	Inhal	Induction/maintenance of anaesthesia
Tiletamine/zolazepam	1–5[7]	IM	Anaesthesia; recovery may be rough
Xylazine	0.5–1.0[6]	IM	Anaesthesia, can give with ketamine
Yohimbine	0.5–1.05	IM	Xylazine reversal

Key: IM=intramuscular, Inhal=inhalation, SC=subcutaneous

[1] (Arnemo and Soli, 1995); [2] (Barbiers, 2003); [3] (Bennett, 2000); [4] (Carpenter, 2005); [5] (Morrisey and Carpenter, 2004); [6] (Smith, 1992); [7] (Smith, 2000); [8] (Stocker, 1992)

Table 6.3: Analgesic doses for hedgehogs

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Buprenorphine	0.01–0.50[1]	SC, IM	6–12	Opioid
Butorphanol	0.05–0.40[2]	SC, IM	6–12	Opioid
Flunixin	0.03[1] 0.3[1]	IM SC	8 24	NSAID

Key: IM = intramuscular, NSAID = non-steroidal anti-inflammatory drug, SC = subcutaneous
[1](Johnson–Delaney, 2002); [2] (Morrisey and Carpenter, 2004)

MARSUPIALS

INTRODUCTION

The sugar glider (*Petaurus breviceps*) is occasionally seen as a pet. The gray (Brazilian) short–tailed opossum (*Monodelphis domestica*) may rarely be presented at veterinary practices.

ANATOMY AND PHYSIOLOGY

Temperature

The metabolism of marsupials is approximately 30% lower than in placental mammals. Basal metabolic rate and body temperature are also lower. Mean body temperatures are between 33°C and 36°C (Holz, 2003).

The optimum environmental temperature for *sugar gliders* is 24–27°C, but they can tolerate a wide range, from 18°C to 32°C (Nagy and Suckling, 1985). During periods of extreme cold, the sugar glider will go into a state of torpor to conserve energy (Fleming, 1980). Heat stress is seen at high temperatures (Ness and Booth, 2004).

Cardio-respiratory disease

Sugar gliders may suffer tachypnoea or dyspnoea due to various aetiologies. Bacterial pneumonia, including *Pasteurella multocida*, and cardiac failure have been reported (Ness and Booth, 2004; Pye and Carpenter, 1999).

Cardiomyopathy and congestive heart failure have been reported in the *grey short-tailed opossum*. Older animals and males appear more susceptible to congestive heart failure (Johnson-Delaney, 2002). The problem should be identified during clinical examination, although confirmatory investigations may require sedation or general anaesthesia. Hyperlipidaemia and hypercholesterolaemia may lead to atherosclerosis in this species.

Urinary system

In marsupials, the cloaca is the combined opening to the urogenital and digestive tracts (Holz, 2003).

Many reports exist of renal disease in *sugar gliders*, including urinary tract obstruction, pyelonephritis/nephritis and renal failure (Johnson-Delaney, 2002) Nephritis, pyelonephritis, and nephrosis have been reported in *grey short-tailed opossums*.

Signs such as polyuria, polydipsia, haematuria and pyuria may indicate renal disease. Clinical assessment before anaesthesia may identify these diseases, but care should be taken not to induce or exacerbate renal problems.

Digestive system

The diet of *sugar gliders* varies seasonally. In the winter, wild animals eat the sugary sap or gum of eucalypts and acacias, nectar from flowers (eucalypts, banksias, acacias and several types of native apple), becoming more insectivorous (eating insects, arachnids and small vertebrates) during the rest of the year. Captive animals should be given a diet that is 50% insectivore diet and 50% fruit sugars, in the form of a sap or nectar. As they are nocturnal, fresh food should be given in the evening (Henry and Suckling, 1984).

Improper diet often predisposes gastrointestinal disease in this species. Animals may be suffering from malnutrition, presenting with hypoproteinaemia, hypocalcaemia, anaemia, and even hepatic and renal abnormalities due to organ damage (Ness and Booth, 2004). They may require nutritional and fluid support before anaesthesia is performed. Other animals may be obese, with cardiac, hepatic or pancreatic disease (Nagy and Suckling, 1985).

The captive *grey short-tailed opossum* is fed pelleted carnivore diet, insectivore diet, or insects, pinky mice and a small amount fruit. Malnutrition and obesity may both be seen in pet grey short-tailed opossums. Neoplasia is frequently found in the liver (Johnson-Delaney, 2002).

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

Hospitalisation facilities

These animals are nocturnal and require quiet surroundings during hospitalisation.

Fluid and nutritional support

It is obviously important to ensure that an appropriate diet is provided during hospitalisation. Both species are nocturnal feeders, so fresh foodstuffs should be offered in the evening (Johnson-Delaney, 2002). Puréed food can be syringefed in small quantities to anorexic patients.

Fluids should be administered during anaesthesia to maintain the renal circulation, particularly in patients where renal disease is suspected. Subcutaneous fluids can be given dorsally in the interscapular region, but they may pool in the patagium (gliding membrane) of sugar gliders. Fluids administered into the patagium will be absorbed very slowly; the intraperitoneal route is more efficacious. If more rapid fluid absorption is required, an intraosseous catheter can be placed (Ness and Booth, 2004).

Fasting

Sugar gliders and opossums require pre-anaesthetic fasting for at least 4 h (Pye and Carpenter, 1999).

TECHNIQUES

Routes of administration

Intravenous access is difficult in these small animals, and only small volumes of drugs may be administered intramuscularly (Table 6.4). The subcutaneous route is suitable for most medications and fluids; large volumes should be pre-warmed.

Table 6.4: Routes of drug administration in sugar gliders and grey short-tailed opossums

ROUTE	COMMENTS
Intramuscular	Epaxial muscles of neck and upper thorax; biceps femoris (anterior thigh)
Intraosseous	Proximal femur
Intraperitoneal	Caudal right abdominal quadrant Collapsed or anaesthetised animals only; useful for fluid therapy
Intravenous:	Difficult; sedation or anaesthesia required Use a 0.5 ml insulin syringe with 27-gauge needle
Cephalic vein	–
Lateral saphenous vein	–
Femoral vein	–
Medial tibial artery	–
Ventral coccygeal or lateral tail vein	Warm tail to dilate tail vessels; tail veins are choice for grey short-tailed opossum
Jugular vein	Ventral neck, halfway between shoulder and ramus of mandible, lateral to trachea and oesophagus (usually general anaesthesia)
Cranial vena cava	Needle inserted at 30° angle just lateral to manubrium, aiming towards contralateral hindlimb (general anaesthesia required)
Cardiac puncture	Emergency use only – may cause fatal haemorrhage
Subcutaneous	Scruff; maximum 10 ml/kg Fluids injected into the sugar glider's patagium will pool and absorption will be slow

(Johnson-Delaney, 2002; Redrobe, 2002)

PRE-ANAESTHETICS

Pre-medication may be administered to provide analgesia, sedation, and/or reduce respiratory and salivary secretions before anaesthesia is induced (Table 6.5).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Drug doses for *sugar gliders* are commonly extrapolated from other species. If metabolic scaling is used, the small body size of these animals is offset by their low metabolic rate (Table 6.6) (Ness and Booth, 2004).

Induction

Volatile agents

The simplest method of inducing anaesthesia in these *small marsupials* is chamber induction with a volatile agent, such as isoflurane or sevoflurane (Fig. 6.1). This is relatively safe even in debilitated animals. Anaesthesia is induced in a chamber with 5% isoflurane.

Table 6.5: Pre-anaesthetic medications in sugar gliders and grey short-tailed opossums

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Atropine	0.01–0.02[4]	SC, IM	Anticholinergic (reduce respiratory and oral secretions)
Diazepam	0.5–1.0[1]	IM	Sedative
Glycopyrrolate	0.01–0.02[3]	SC, IM, IV	Anticholinergic (reduce respiratory and oral secretions)
Ketamine	20[2]	IM	Sedation before induction/maintenance with isoflurane (sugar glider)

Key: IM = intramuscular, IV = intravenous, SC = subcutaneous
[1] (Barnes, 2002); [2] (Hough et al., 1992); [3] (Johnson–Delaney, 2000); [4] (Morrisey and Carpenter, 2004)



Figure 6.1 • Chamber induction of anaesthesia using isoflurane in a sugar glider, *Petaurus breviceps*.

Injectable agents

Tiletamine/zolazepam has been used to anaesthetise sugar gliders, at 8.4–12.8 mg/kg (Bush et al., 1990). However, some animals suffered neurological signs and death (Holtz, 1992).

Anaesthetic maintenance

Anaesthesia is usually maintained using 2–3% isoflurane via a closely fitting facemask. It may be possible to intubate the sugar glider using a stylet and fine-bladed laryngoscope (Booth, 2000). The head is extended and the tongue gently grasped in order to pass a 1 mm endotracheal tube into the larynx (Cook, Global Veterinary Products, New Buffalo, MI) (Ness and Booth, 2004).

Recovery

A warm quiet area should be provided for recovery. Supplemental oxygen should be provided until the patient is making coordinated movements.

Table 6.6: Anaesthetic and analgesic drugs in sugar gliders

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Acepromazine + butorphanol	1.7 + 1.7[2]	PO	Postoperative sedation and analgesia to prevent self-trauma at incision site
Acepromazine + ketamine	1 + 10[2]	SC	Postoperative sedation and analgesia to prevent self-trauma at incision site
Butorphanol	0.5[6]	IM	Analgesia; can repeat q8h
Flunixin	0.1–1.0[3,4]	IM	Analgesia; repeat q12–24h, use for up to 3 days
Isoflurane	5%[1] 1–3%[7]	Inhal	Anaesthetic of choice Induction Maintenance
Sevoflurane	To effect[5]	Inhal	Anaesthesia

Key: IM=intramuscular, Inhal=inhalation, PO=oral, q8h=every 8 hours, SC=subcutaneous
[1] (Barnes, 2002); [2] (Johnson, 1997); [3] (Johnson–Delaney, 2002); [4] (MacPherson, 1997); [5] (Morrisey and Carpenter, 2004); [6] (Ness, 2000); [7] (Pye, 2001)

ANAESTHESIA MONITORING

Cardio-respiratory parameters and reflexes are similar to other small mammals.

The cloacal temperature is lower than body temperature; rectal temperature is more closely allied to core body temperature. To reach the rectum, the *thermometer* should be directed dorsally from the cloaca (Fleming, 1980). Core temperature can also be measured at the tympanic membrane (Ness and Booth, 2004).

PERI-ANAESTHETIC SUPPORTIVE CARE

Supportive *heating* is necessary to prevent hypothermia during anaesthesia. Heat pads and forced warm-air blankets are useful during anaesthesia, with recovery in an incubator (Ness and Booth, 2004).

Analgesia

For painful conditions or surgery, analgesia should be administered routinely (Table 6.6). Sugar gliders are particularly adept at self-traumatising wounds, and further analgesia may be required post anaesthesia (sometimes in combination with a sedative, for example 1.7 mg/kg butorphanol with 1.7 mg/kg acepromazine diluted with normal saline and administered orally (Johnson, 1997)).

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7 Non-human primate anaesthesia

INTRODUCTION

The most popular pet primate is the common marmoset, *Callithrix jacchus*. Other callitrichids (marmosets and tamarins), capuchins and squirrel monkeys may also be seen. Legal requirements may necessitate a licence or other form of monitoring these animals in captivity. It should be borne in mind that non-human primates may transfer zoonotic infections to human handlers, but conversely they will be susceptible to many human-borne diseases. Latex or nitrile gloves should be worn to protect both staff and pet, along with respiratory protection (for example, disposable facemask) if either has a respiratory disease. Herpes simplex virus causes cold sores in humans, but may cause a fatal infection in callitrichids (marmosets and tamarins) (Thornton, 2002).

ANATOMY AND PHYSIOLOGY

Temperature

Primates are fairly adaptable to a wide range of temperatures. Ambient enclosure temperature should be above 15°C, and up to 29°C in a basking area (Thornton, 2002).

Cardiovascular system

Pet primates on inappropriate diets can be obese. This may be associated with cardio-respiratory disease, as seen in humans.

Respiratory system

Measles is a differential diagnosis for animals with a mucoid nasal discharge, and this virus may also cause interstitial and bronchopneumonia. Other aetiological agents of pneumonia in primates are *Bordetella bronchiseptica*, *Klebsiella pneumoniae* and *Pasteurella* spp. *Toxoplasma gondii* can cause pulmonary oedema (Thornton, 2002). Sedation or anaesthesia may be required for investigation of respiratory disease and identification of aetiological agents, but extra care should be taken with these patients who will be at greater risk during sedation or anaesthesia.

Digestive system

Non-human primates are omnivorous. Their diets vary between species, ranging from fruits, gums and saps, to invertebrates and small vertebrates. Captive animals may be fed pellets, and/or gum substitutes. The diet of the common marmoset is mainly fruit.

Primates have a daily requirement for vitamin C. New World primates require vitamin D_3, particularly callitrichids (marmosets and tamarins), and require an ultraviolet light (UV-B) for calcium and phosphorus metabolism. Invertebrates can be gut-loaded with calcium supplement.

Dietary protein requirements are high in primates. Mice should not be fed to callitrichids, as they are susceptible to lymphocytic choriomeningitis virus (which is carried by mice and causes fatal callitrichid hepatitis).

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

Assessment before anaesthesia is important in all species. However, these animals are potentially dangerous and many will bite when restrained. The risk to staff handling animals that are not tamed can be significant, not only from bite and scratch injuries, but also from disease transmission.

Smaller species, such as common marmosets, can usually be caught with a towel or leather gloves for assessment and anaesthetic induction (Fig. 7.1). Larger species may require the administration of injectable anaesthetic drugs to induce anaesthesia while restrained in a squeeze cage before a 'hands-on' examination can be performed.



Figure 7.1 • Mask induction with isoflurane of a common marmoset (*Callithrix jacchus*).



Figure 7.2 • Intravenous catheter in the cephalic vein of a brown capuchin (*Cebus apella*).

EQUIPMENT REQUIRED

Due to the risks of disease transmission between non-human primates and staff, it is advisable to wear personal protective equipment, such as examination gloves and facemasks. It is preferable not to have ill personnel, for example those with respiratory disease, dealing with non-human primates.

TECHNIQUES

Routes of administration

Intravenous access (Fig. 7.2) is more difficult in smaller species, but most routes are accessible for drug and fluid administration (Table 7.1).

Intubation

It is advisable to intubate primates when anaesthetised, to protect their airway and to provide oxygen. A laryngoscope is useful to aid visualisation of the larynx, particularly in larger species (Fig. 7.3). Local anaesthetic should be sprayed on the larynx to reduce the risk of laryngospasm. Short endotracheal tubes are used, to avoid obstructing one bronchus whilst intubating the other at the proximal tracheal bifurcation (Thornton, 2002). A 2–2.5 mm diameter endotracheal tube can be used in primates weighing approximately 500 g (for example, common marmosets). Positioning can be assessed by auscultating the lungs to check for bilateral ventilation.

PRE-ANAESTHETICS

Acepromazine is used as a pre-medication rarely, its main advantage being the possibility of oral administration. Similarly, oral diazepam or chlorpromazine will cause sedation.

Atropine may be administered to reduce oral and respiratory secretions (at 0.02–0.05 mg/kg subcutaneously, intramuscularly or intravenously) (Holmes, 1984). Glycopyrrolate has also been used in larger non-human primate species (<0.01 mg/kg) (Popilskis and Kohn, 1997).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Volatile agents

Small primates such as marmosets are easiest induced using volatile agents in a chamber or restrained for facemask induction. The use of agents such as isoflurane carries a low risk

Table 7.1: Routes of administration in non-human primates

ROUTE	TECHNIQUE	COMMENTS
Intramuscular	Quadriceps muscles, lumbar muscles	Small volumes only
Intraosseous	Proximal femur, proximal tibia	Useful in collapsed or small animals when intravenous access not possible. Provide analgesia
Intraperitoneal	Caudal right abdominal quadrant	In collapsed or anaesthetised animals only
Intravenous:		Sedation or anaesthesia usually required
Femoral vein	Medial to femoral artery, which can be palpated 25 or 27 gauge needle, 1–2 ml syringe	Apply pressure for a few minutes after venepuncture to avoid haematoma formation
Lateral saphenous Jugular vein Cephalic vein		Lateral saphenous, jugular and cephalic very difficult to access in small species
Subcutaneous	Scruff, lateral body wall	Maximum volume 15–20 ml in common marmoset, 125–190 ml in capuchin

(Thornton 2002; Joslin 2003)

of morbidity and mortality, and is, therefore, preferable for patients with respiratory disease or other debilitation. Isoflurane is also commonly used after induction with injectable agents, either to deepen or prolong anaesthesia.

Halothane, with a minimum alveolar concentration (MAC) of 0.98–1.15%, produces dose-dependent cardiovascular depression (Steffey et al., 1974). Isoflurane produced respiratory depression when administered with morphine, but no cardiovascular depression (Steffey et al., 1994). The MAC for isoflurane is 1.28–1.46%. Sevoflurane has an MAC of 2%. One study reported no significant cardiopulmonary effects during anaesthesia with this agent (Soma et al., 1988). Changes were, however, noted in body temperature (lowered), and haematology and clinical chemistries (lower white cell count, calcium, and total protein).

Nitrous oxide has an MAC of 200% in non-human primates (Popilskis and Kohn, 1997). It is used to provide a 'second gas effect', reducing the concentrations of other volatile agents required.



Figure 7.3 • Anaesthesia was induced in this brown capuchin (*Cebus apella*) with intramuscular medetomidine and ketamine. The animal is being intubated after local anaesthetic has been applied to the larynx to reduce laryngospasm, and will be maintained on isoflurane.

Injectable agents

These are particularly useful for animals that cannot be safely removed from cages, such as larger species that are not routinely handled. Unfortunately, there is great variation in response to anaesthetic agents between species.

Most anaesthetic combinations for non-human primates contain *ketamine*, which has a wide safety margin when used in non-human primates. Ketamine on its own will cause sufficient immobilisation for handling and clinical examination, but muscle relaxation is poor. Lemurs may have a seizure if ketamine is used as the sole agent, but can be anaesthetised with combinations of the drug. The use of combinations allows lower doses of ketamine to be utilised, reducing the side effects and recovery time. Combinations commonly used include ketamine with medetomidine or diazepam; both will cause anaesthesia.

Side effects seen with medetomidine include bradycardia, hypotension, loss of thermoregulation, and decreased mean arterial pressure (Capuano et al., 1999). On induction with medetomidine, a transient increase in respiratory rate is seen, followed by a more prolonged decrease. Xylazine can be used with ketamine, but often leads to vomiting on induction. Medetomidine and xylazine can be reversed with atipamezole. Diazepam or midazolam causes less cardio-respiratory depression, but reversal of the benzodiazepines with flumazenil is expensive.

Similar agents are found in the *tiletamine* (another dissociative anaesthetic) and *zolazepam* (a benzodiazepine) combination. Different species require a wide range of doses. Side effects may include severe hypothermia

Table 7.2: Sedative and anaesthetics for primates

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Acepromazine	0.5–1.0[10]	PO, SC, IM	Pre-anaesthetic, tranquillisation
Alfaxalone/ alphadalone		IM, IV	Higher doses required IM
			Rapid induction and recovery
	5–8[6,16]		Light sedation
	12–18[16]		Deep sedation, light anaesthesia
	18–25[12,16]		Surgical anaesthesia
	10 mg/kg/h[6]		Continuous rate infusion
Atipamezole	4 × medetomidine dose[2]	SC, IM, IV	Specific alpha-2-antagonist, used for medetomidine (or xylazine) reversal
Chlorpromazine	1–6[10]	PO, IM	Pre-anaesthetic
Diazepam			Sedation after 30–60 min, variable degree of effects
	0.5–1.0[7]	PO	
	0.25–0.50[9]	IM, IV	Prolonged recovery
Fentanyl	10–25 μg/kg/h[13]	IV	Continuous infusion Isoflurane-sparing
Fentanyl/droperidol (Innovar-Vet®, Janssen)	0.05–0.10 ml/kg[10] 0.1–0.3 ml/kg[13]	IM, IV SC, IM	Pre-anaesthetic; respiratory depression at high doses Minor procedures
Fentanyl/fluanisone (Hypnorm®, Janssen)	0.3 ml/kg[13]	SC, IM	Minor procedures
Flumazenil	0.025[8]	IV	Benzodiazepine reversal
Halothane	0.5–1.0 %[15]	Inhal	Can supplement with nitrous (2:1 with oxygen) during maintenance
Isoflurane		Inhal	Preferable to pre-medicate with injectable agent
	4%[16]		Induction
	0–3%[16]		Maintenance
Ketamine	5–20[3,7]	IM	Immobilisation for handling or examination; induce with volatile agent Use lower dose with larger animals (e.g. 20 mg/kg for callitrichids, such as marmosets and tamarins, 5 mg/kg for great apes) Causes seizures in lemurs
Ketamine + diazepam	15 + 1[4,16]	IM	Surgical anaesthesia with good muscle relaxation

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Ketamine + medetomidine	10.0–15.0 + 0.1[16]	IM	Light anaesthesia Top up with isoflurane or half dose of ketamine Lower dose in large primates
Ketamine + midazolam	15 + 0.05–0.09[13] 15 + 0.05–0.15[13]	IM + IV	Animal <1 kg body weight Animal >1 kg weight
Ketamine + xylazine	10.0 + 0.5[4]	IM	Often vomit on induction Surgical anaesthesia
Medetomidine	0.01–0.1[1,11]	PO, IV	Sedation/induction Many side effects (see text)
Midazolam	0.1–0.5[7,13]	IM, IV	Sedation Often used in combination with fentanyl (1–2 μg/kg IV)
Nitrous oxide	<30%[15,17]	Inhal	Reduces concentration of other volatile agents required
Propofol	5–10[16]	IV	Apnoea common. Can give continuous infusion
Sevoflurane	8%[14] 2.5 %[14]	Inhal	Induction Maintenance
Thiopentone	15–20[5]	IV	Surgical anaesthesia Halve dose if ketamine pre-medicant
Tiletamine + zolazepam	1–10[7,16]	IM	Wide species variation in dose required

Key: Inhal = inhalation, IM = intramuscular, IV = intravenous, PO = orally, SC = subcutaneous

[1] (Capuano et al., 1999); [2] (Carpenter, 2005); [3] (Eberhard, 1982); [4] (Flecknell, 1987); [5] (Flecknell, 1996); [6] (Foster et al., 1996); [7] (Heard, 1993); [8] (Horne et al., 1998); [9] (Ialeggio, 1989); [10] (Johnson et al., 1981); [11] (Kearns et al., 1998); [12] (Phillips and Grist, 1975); [13] (Popilskis and Kohn, 1997); [14] (Soma et al., 1988); [15] (Steffey et al., 1974); [16] (Thornton, 2002); [17] (Tinker et al., 1977)

(macaques) or ataxia during recovery (great apes) (Horne, 2001; Lopez et al., 2002).

Propofol can be used in primates, but the need for intravenous administration limits its usefulness. Side effects include a reduction in mean arterial pressure, heart rate and myocardial contractility (Fanton et al., 2000). *Alfaxalone/alphadolone* will cause dose-dependent sedation or anaesthesia, but large volumes are required via intramuscular injection.

Fentanyl has been shown to have a sparing effect on isoflurane. It may be administered by bolus or continuous infusion with isoflurane anaesthesia. Fentanyl may also be given in combination with midazolam.

Other drugs and combinations have been used to cause sedation or induce anaesthesia in non-human primates (Table 7.2). The use of neuromuscular blocking agents, such as succinylcholine, atracurium, pancuronium, tubocurarine and vecuronium, is reported in non-human primates. Assisted ventilation is required if they have been administered. Succinylcholine has a rapid onset and short duration of action. The other four listed are non-depolarising neuromuscular blockers and are reversed

BOX 7.1 Anaesthesia induction in non-human primates

- Ketamine is well tolerated, and is often used in combinations for anaesthesia of non-human primates
- Volatile agents, such as isoflurane, cause rapid induction and recovery from anaesthesia. This is the agent of choice in debilitated animals

using neostigmine, usually in combination with an anticholinergic agent to reduce the bradycardia that neostigmine induces. The use of neuromuscular blocking agents in pet species is seldom indicated.

Anaesthetic maintenance

Volatile agents are commonly used to top up or maintain anaesthesia. If possible, the patient is intubated (Fig. 7.3). If this is not feasible, a close-fitting facemask is used to administer oxygen and anaesthetic gases.

Recovery

Oxygen should be supplemented via facemask until the patient is showing signs of voluntary movement. To avoid staff injuries (for example bites), the primate is gently but firmly restrained once volatile agents have been switched off and/or reversal agents injected. Supplemental heating should be continued until the patient is moving around in a coordinated manner.

Suggested anaesthetic protocols

Most small primates kept as pets can be induced in a chamber. This reduces the stress of restraint to the animal and reduces the risk of staff injury. Habituated animals may be held during mask induction (Fig. 7.1). After induction, it is preferable to intubate the patient and maintain on volatile agents.

In larger animals, ketamine and medetomidine are commonly used to induce anaesthesia, which may be topped up as required with volatile agents using a facemask or via an endotracheal tube if they have been intubated (Fig. 7.3). Recovery is more prolonged with injectable agents. Medetomidine can be reversed with atipamezole and benzodiazepines (diazepam and midazolam) with flumazenil.

ANAESTHESIA MONITORING

Cardio-respiratory monitoring is as for other small mammals. Heart and respiratory rates should be assessed throughout the procedure using a bell or oesophageal *stethoscope*. A rectal *thermometer* can be used to monitor body temperature.



Figure 7.4 • This common squirrel monkey, *Saimiri sciureus*, has been induced with medetomidine and ketamine before intubation and maintenance with isoflurane. Echocardiogram pads are attached to the feet to monitor electrical activity of the heart, and a pulse oximeter on the ear is used to assess arterial oxygen saturation. A warm air blanket is being used to provide supplemental heating during anaesthesia.

Electrocardiogram (ECG) leads can be attached to the feet using pads (Fig. 7.4) or clips used as in larger species. *Pulse oximeters* can be attached to the tongue or ear.

PERI-ANAESTHETIC SUPPORTIVE CARE

Supplemental heat is important during anaesthesia and recovery, particularly for small individuals. Analgesia should be given if a painful procedure is to be or has been performed or the animal is perceived to be in discomfort. The recovery cage should be on one level, to avoid animals climbing and injuring themselves if they fall. A soft substrate should be provided, for example towelling or straw. Animals that are part of a social group should be returned to the group as soon as possible after recovery; otherwise there may be problems with re-acceptance into the group.

BOX 7.2 Group-housed non-human primates

- Animals should be fully conscious before returning to their group to avoid being traumatised by companions.
- The total time apart from the group (including the hospitalisation period pre-anaesthesia) should be minimised to avoid re-acceptance problems.

Analgesia

The administration of anti-inflammatories and analgesia to non-human primates is particularly important after surgery, as these species are adept at removing sutures or traumatising lesions (Table 7.3).

Table 7.3: Analgesics in primates

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Aspirin (acetylsalicylic acid)	5–10[11]	PO	4–6	–
Bupivacaine	1[8]	Local	–	0.25 %; e.g. intercostals nerve block
	1.2[3]	Epidural	–	Bupivacaine hydrochloride, 0.5%
Buprenorphine	0.01[4]	IM, IV	12	–
Butorphanol	0.1–0.2[4]	IM	12–48	μ receptor agonist in primates May cause profound respiratory depression
Carprofen	2–4[9,11]	PO, SC	12–24	Differs with species so calculate frequency to effect
Fentanyl	4–8 μg/kg/h[1]	Transdermal patch	–	Monitor for respiratory depression
Flunixin	0.3–1.0[4]	SC, IV	12–24	Care in dehydrated patients
Ibuprofen	20[7]	PO	24	–
Ketoprofen	5[9]	IM	6–8	–
Morphine	1–2[10]	PO, SC, IM, IV	4	Dose-dependent respiratory depression. May cause facial pruritus[5]
	0.01[8]	Intrathecal	–	Preservative-free morphine
	0.1[8]	Epidural	–	Preservative-free morphine
Nalbuphine	0.5[4]	IM, IV	3–6	Agonist-antagonist opioid
Oxymorphone	0.03–0.2[4]	SC, IM, IV	4–12	Use low dose in New World primates, higher dose in Old World primates[6]
Paracetamol	5–10[11]	PO	6	–
Pethidine	2–4[2]	IM	3–4	–

Key: IM = intramuscular, IV = intravenous, PO = orally, SC = subcutaneous

[1] (Carpenter, 2005); [2] (Flecknell, 1987); [3] (Golub and Germann, 1998); [4] (Heard, 1993); [5] (Horne, 2001); [6] (Isaza et al., 1992); [7] (Patton et al., 1997); [8] (Popilskis and Kohn, 1997); [9] (Ramer et al., 1998); [10] (Rosenburg, 1991); [11] (Thornton, 2002)

EMERGENCY DRUGS

Table 7.4: Emergency drugs in non-human primates

DRUG	DOSE (mg/kg)	ROUTE	INDICATION/COMMENT
Adrenaline (epinephrine)	0.2–0.4 (diluted in 5 ml sterile water)[2] 0.5–1.0 (of 1:10 000 dilution)[2]	IT IV	Cardiac arrest
Atropine	0.02–0.05[5]	IM	Anticholinergic – bradycardia
Diazepam	0.25–0.50[3]	IM, IV	Seizures
Doxapram	2[1]	IV	Respiratory stimulant
Frusemide	1–2[2]	IV	Duresis; heart failure; pulmonary oedema
Glycopyrrolate	0.005–0.010[5]	IM	Anticholinergic
Lidocaine (lignocaine)	1–2[2]	IV	Ventricular arrhythmias; premature ventricular contractions
Prednisolone sodium succinate	1–15[4]	IV	Shock
Naloxone	0.01–0.05[1]	IM, IV	Opioid reversal

Key: IM = intramuscular, IT = intratracheal, IV = intravenous

[1] (Flecknell, 1987); [2] (Fortman et al., 2002); [3] (Ialeggio, 1989); [4] (Melby and Altman, 1976); [5] (Popilskis and Kohn, 1997)

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Fancy pigs anaesthesia

8

INTRODUCTION

The ancestor of domesticated pigs is the wild pig (*Sus scrofa*). Many breeds have now been developed, with the most common being the large production species. Pet pigs are infrequently presented to the veterinary practitioner. This chapter will cover the basics of pig anaesthesia as it pertains to those breeds kept as pets or on 'hobby' farms. Although the Vietnamese Potbellied pig is a well-known and popular pet breed, veterinary clinicians in general practice may also be presented with rare breeds, such as the Tamworth (Fig. 8.1), Berkshire (Fig. 8.2) or Kune Kune (Fig. 8.3). Most animals are kept outdoors in a pen with a shelter, but some are house-trained. Many will be kept singly, but some owners will have a small group.

The adult Potbellied pig usually weighs 50–70 kg, but is prone to obesity and may weigh more. They are descendents of the Banded pig in South-East Asia. Another popular breed is the Göttingen Minipig, which weighs 35–45 kg. These were developed in the 1960s by crossing Minnesota Minipigs with Vietnamese Potbellied pigs. This breed is commonly used in laboratories for experimental purposes (Sambraus, 1992). The Kune Kune originates from New Zealand.

As pet pigs are susceptible to infectious diseases seen in production breeds, they may be subject to similar restrictions. Legislation may place movement or housing restrictions to minimise the spread of disease, for example foot and mouth disease and swine fever.

Manual restraint is difficult in all ages of pig, and sedation or anaesthesia may be required for a wide range of procedures, from phlebotomy to surgery. Many of the risks associated with anaesthetising the previously discussed small mammals do not apply to pigs, mainly due to their larger size, and considerations are more akin to those in dogs. However, the pig's unique anatomy will present the veterinary practitioner with some specific problems.



Figure 8.1 • Tamworth (rare breed often kept as a pet).

ANATOMY AND PHYSIOLOGY

Skull shape varies between pig breeds, with those such as the Middle White (Fig. 8.4) having quite convex facial features. The snout is quite short in Potbellied pigs. The short neck is not distinct and subcutaneous fat blends the outline of the head into that of the body (Dyce et al., 1987).

Curved canines project from the mouth. In boars these tusks grow throughout life, but stop growth after 2 years in



Figure 8.2 • Berkshire (rare breed often kept as a pet).



Figure 8.3 • Kune Kune (rare breed often kept as a pet).

sows (Dyce et al., 1987). Care should be taken in conscious or sedated animals to avoid injury to staff or owners. Conscious animals may bite, particularly when stressed during restraint. Most animals are vociferous when restrained and people in the vicinity should wear ear defenders to avoid hearing damage.

Temperature

Temperature requirements vary greatly, depending mostly on the size of animal. Heavy animals are able to live in cooler environmental temperatures, but young and small animals require a higher ambient temperature (Taylor, 1995).

Cardiovascular system

The heart is relatively small in the pig, extending from the second to fifth ribs in the ventral thorax. Heart rate is usually 70–80 beats per minute, but can be up to 280 beats per minute in newborn piglets (Taylor, 1995).



Figure 8.4 • The Middle White has a convex dorsal skull shape.

Many sites exist for venepuncture in pigs, but difficulties exist with all. Unless sedated or anaesthetised, most pigs will object both physically and vociferously to venepuncture. The most accessible and superficial vein is the auricular vein. The use of local anaesthetic ointment (for example, EMLA®, AstraZeneca, London, UK) and restraint may allow venepuncture in smaller animals, but chemical immobilisation to some degree will be necessary for most patients.

The jugular vein is not visible in pigs, but lies in the region of the jugular furrow (which may be covered by subcutaneous fat in larger specimens). In anaesthetised animals, a cutdown procedure is often required to access the jugular vein for catheterisation. Similarly, the femoral vein is accessible in anaesthetised animals. A more common site for phlebotomy is the anterior vena cava. If possible, the animal is restrained in a bleeding crate (Taylor, 1995). In an emergency situation, with collapsed or anaesthetised animals, intracardiac injection is possible. Good injection technique is important, as porcine blood vessels are very fragile and will tear easily or spasm when injected. Most areas of skin are relatively hairless so do not require clipping, but the area should be prepared aseptically before needle insertion.

Respiratory system

The resting respiratory rate varies greatly depending on the animal's age (Table 8.1). The average in adults is 20–30

Table 8.1: Basic physiological data of pigs	
Body temperature (°C)	
Higher temperature in newborns, lower in adults	39 (38.4–40)
Respiratory rate at rest (per minute)	
Young	<50
Adult	20–30
Old sows	13–15
Heart rate at rest (per minute)	
Newborn	200–280
Adult	70–80
Minimum environmental temperature requirements (°C)	
Piglets with sow (<5 kg)	25–30
Piglets 3–6 weeks old (alone)	27–32
Weaned 6–12 weeks old	21–24
Growing 12–16 weeks old	15–21
Pigs 16–26 weeks old	13–18
Adult in house/yard	15–20
Adult at farrowing	15–18
Maximum environmental temperature (°C)	
Adult in house/yard	28

(Straw and Merten, 1992; Taylor, 2002)

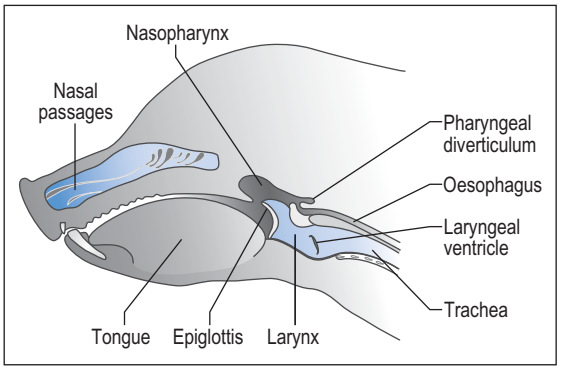


Figure 8.5 • Schematic of upper respiratory tract (sagittal section through skull).

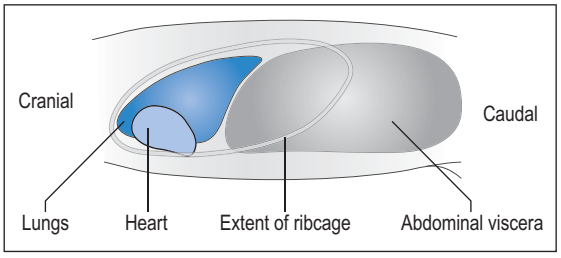


Figure 8.6 • The areas occupied by the lungs and the heart are relatively small in pigs.

breaths per minute, but is up to 50 breaths per minute in newborn piglets, and as low as 13–15 in old animals (Taylor, 1995).

The nasal cavities are narrow. The middle and ventral meatuses carry air from the nares via the choana to the nasopharynx (Fig. 8.5). In animals suffering from rhinitis, the nasal passages will be even narrower and may become deformed (Dyce et al., 1987). This will affect the provision of oxygen to anaesthetised patients via nasal catheters.

Although the lips are long, the mouth does not open very wide. Visualisation of the caudal oral cavity and larynx for intubation is, therefore, difficult. The soft palate is long and the tongue relatively fatty. The porcine laryngeal anatomy produces various obstacles to intubation. The pronounced arytenoids and ventrally sloping thyroid cartilage make entry of the larynx difficult. The long thyrocricoid ligament on the ventral larynx presents a blind-ending pouch that must also be avoided during intubation. There is also a pharyngeal recess dorsal to the larynx and a laryngeal recess in the midline between the base of the epiglottis and thyroid cartilage on the ventral larynx (Hodgkinson, 2007). The trachea follows from the larynx dorsally at an acute angle (Fig. 8.5) (Dyce et al., 1987).

The lungs do not project far caudally in pigs, covering only the cranial third to half of the thorax (Fig. 8.6) (Braun and Casteel, 1993). Auscultation and percussion of lungfields are made more difficult by subcutaneous fat and variable patient demeanour.

Although infectious aetiologies commonly cause pneumonia in pigs, disease is often predisposed by poor husbandry (see Pre-anaesthetic section).

Digestive system

Commercial pig diets or Potbellied pig special diets (for example, Mazuri Porcine Mini-Pig Diets®, Purina Mills) are available. Table scraps and treats should be avoided, as obesity is very common in Potbellied pigs and contact with meat may allow transmission of various diseases (Braun and Casteel, 1993; Taylor, 2002). Homemade diets may cause malnutrition and hypothermia may be predisposed by a low-energy diet.

Diarrhoeas are often infectious and may include zoonotic diseases, such as *Salmonella*. Besides supportive care and fluids for dehydrated animals, isolation and hygiene are paramount for patient care.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History

Details of the animal's history and husbandry may elucidate factors that predispose to disease. For example, poor ventilation and unclean bedding may allow ammonia levels to build up that will be irritant to respiratory surfaces, predisposing to infection. Dampness, draughts and overcrowding will also contribute to disease development (Braun and Casteel, 1993). The demeanour of pigs is variable – a full clinical examination may be possible in habitualised animals, whilst only a cursory examination is possible in others. Pig-boards and the judicious use of a snare on the maxilla may be required to restrain the patient for examination and administration of drugs. The owner should be warned that restraint will usually result in vocalisations.

Clinical assessment

Initial observations should include assessment of the animal's demeanour, locomotion, and respiratory rate and pattern. Nasal discharges may be seen with respiratory tract disease. Thoracic auscultation should be attempted, but may be unreliable in detection of lung pathology due to the difficulties associated with the procedure (see Anatomy section). The heart can be auscultated in amenable animals. The skin is easily visible due to the relative hairlessness in most breeds, and skin colour changes due to disease or haemorrhages will be seen on pale-coloured animals. Abdominal palpation is unrewarding in larger pigs, but a superficial examination is possible. A rectal temperature may be taken. Rectal examination is difficult in conscious animals. The external genitalia and mammary glands can be visualised and palpated for abnormalities. The limbs can be palpated for abnormalities and potential sites for intravenous access identified.

An accurate weight should be obtained if possible and is particularly important if injectable anaesthetic agents are to be used. In piglets, small animal digital scales or a spring balance with a sack or bucket may be used. In larger animals, scales for farm animals will be more appropriate.

Fluids and nutritional support

Owing to the difficulties associated with conscious restraint in pigs, supportive care may be limited in this species. Oral supplements can be offered to animals that are still self-feeding and drinking. Colostrum or sow milk substitutes are available for piglets and can be administered via gavage tube.

Fluids should be administered during anaesthesia, particularly if a prolonged procedure is anticipated. Intravenous administration is optimal, but in small patients warmed fluids can be administered intraperitoneally as in other species. To reduce the risk of hypothermia, fluids should be warmed to 38°C for neonatal or small patients (Hodgkinson, 2007).

A balanced electrolyte solution such as Hartmann's is commonly administered intravenously at 10 ml/kg/h (Hodgkinson, 2007). Alternatively, a bolus of 500 ml Hetastarch 10% (for an average 40 kg pig) can be followed by a continuous rate infusion of Ringer's lactate solution at 3–5 ml/kg/h (Goldman et al., 2005).

Fasting

Pigs rarely vomit on induction of anaesthesia, but should be fasted for 6–12 h prior to anaesthesia to reduce the possibility of vomition and aspiration (Flecknell, 1996; Hodgkinson, 2007). This will also reduce the pressure from the stomach on the diaphragm that can reduce respiratory efficiency. If abdominal surgery is to be performed, the pig should be fasted for 12–24 h. Neonates and paediatric patients should not be fasted due to the risk of hypoglycaemia. Water should be removed 4 h before induction (Braun and Casteel, 1993).

EQUIPMENT REQUIRED

Pigs are usually vociferous when restrained, with the noise produced sufficient to cause damage to the human auditory system. It is, therefore, advisable to warn the owner that the animal will squeal and to wear ear protectors.

Owing to the relatively narrow tracheal diameter and acute angle between oropharnyx and larynx, relatively small *endotracheal tubes* are required for porcine intubation (from 3.5 mm for a 5 kg animal to 16 mm for a large adult). A long straight laryngoscope blade is necessary to aid visualisation of the larynx. Laryngeal mask airways are an alternative to endotracheal intubation, or soft tubing can be used as nasal catheters (Goldman et al., 2005).

Anaesthetic circuits will depend on the size of patient. Non-rebreathing circuits used in small animal veterinary practice are suitable for most minipigs, using a T-piece or Bain for young pigs. For larger patients, a rebreathing system will be more efficient, for example a small animal circle for those weighing over 15 kg. A large animal circle will be necessary for patients weighing more than 135 kg (Hodgkinson, 2007).

Monitoring equipment should include an oesophageal stethoscope, digital rectal thermometer (or thermistor probe) and pulse oximeter.

RELEVANT TECHNIQUES

Routes of administration

Drug administration in pigs can be difficult. All animals will vocalise when restrained or injected. Adults usually have a deep layer of subcutaneous fat, with an associated poor blood supply and poor absorption.

Oral medications can be given with food. Direct oral administration can be dangerous in all but the smallest of patients due to the presence of large sharp canines.

Subcutaneous injections can be administered just caudal to the base of the ear, avoiding the parotid gland that lies ventral to the ear (Dyce et al., 1987).

Intramuscular injections are administered just caudal to the base of ear, into the muscle mass attaching to the caudal skull surface. The neck muscle here is the preferred site, as it has relatively little subcutaneous fat and is well tolerated by most pigs (Hodgkinson, 2007). A needle up to 25 mm long for smaller pigs or 38 mm for adult pigs should be used to ensure the injection is deeper than the subcutaneous fat layer (Braun and Casteel, 1993).

Intravenous access can be difficult in pigs, as many animals are not routinely handled and resist restraint.

The *anterior vena cava* is often used for phlebotomy as a large sample can be obtained. This may be easier with the pig in dorsal recumbency. The animal is restrained with the neck in extension and the forelimbs pulled straight in extension away from the head (as for cat jugular venepuncture). A needle (>32 mm long for a large adult) is inserted into the thoracic inlet (between the manubrium sterni and shoulder joint) in a caudal direction, 45° to vertical, to a depth of 2–3 cm. The right side is preferred, as the left phrenic nerve and unpaired thoracic duct (which lies more to the left) may be damaged. Positioning is checked by aspirating blood. This vessel cannot be catheterised.

Other sites, such as the right brachiocephalic and right external jugular vein, may be accessed in anaesthetised patients. To access the *jugular vein*, the neck is held in extension with the forelimbs straight as for anterior vena cava puncture. Pressure is applied at the thoracic inlet to raise the vein. The needle is inserted at the level of the shoulder joint in the region of the jugular furrow (which may not be visible in fatter animals). The vein lies relatively deep in the tissues of the neck (beneath the skin, fat and muscles) and jugular venepuncture is possible only in smaller animals without excess subcutaneous neck fat. Jugular catheterisation requires a surgical cut-down technique under anaesthesia.

The *auricular vein* (Fig. 8.7) is the easiest superficial vein to access, but catheterisation will be difficult unless the animal is sedated or debilitated. This vein can be injected into or catheterised from the caudal surface of the pinna. In conscious animals, local anaesthetic cream (lidocaine/prilocaine, EMLA®, AstraZeneca, London, UK) should be applied 30 min before venous access is required, to reduce the discomfort of venepuncture and resultant movements by the patient. If the animal is conscious, a snare on the snout may be used for restraint. The skin of the pinna is relatively hairless in most patients and clipping is not usually required, although the skin should be cleaned with antiseptic before the vein is entered. Manual pressure or a tourniquet at the base of the ear will raise the vein. A butterfly catheter (22–25-gauge) with extension tubing is useful in case of patient movement; alternatively an intravenous catheter can be placed.

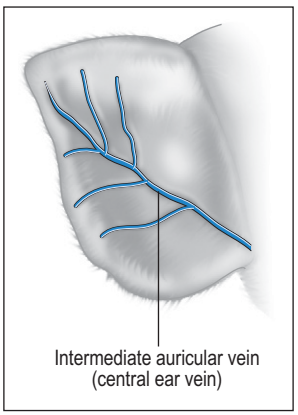


Figure 8.7 • Diagram of the caudal surface of a pig's ear, demonstrating the auricular vein for catheterisation.

The *cephalic vein* is accessible in most anaesthetised animals, but cut down may be required through the thick skin and it is difficult to access in animals with short fat legs. This is a good site for catheterisation and fluid administration during anaesthesia. Access is also difficult in larger, older animals with thickened skin over the antebrachium. A cut-down technique may be required and it is usually only possible in anaesthetised or collapsed animals. The *lateral saphenous vein* may be used similarly in anaesthetised animals, but is difficult in Potbellied pigs due to their skin folds.

The *subcutaneous abdominal vein* is usually easy to visualise and access (including catheterisation) in porcine species, even in conscious animals (Snook, 2001).

Intracardiac injection may be used in an emergency (see Fig. 8.6). The heart is located between the second and fifth ribs. It is entered 2–3 cm cranial and 2 cm dorsal to the apex beat on the left-hand side, via the left fifth or right fourth intercostal space. Access is easier from the left-hand side as there is more contact between the heart and body wall than on the right. Pulling the forelimb forward will enhance access.

The *orbital sinus* can be accessed for small blood samples in anaesthetised pigs. There is a risk of ocular damage with this technique. A 20-gauge 25 mm needle is inserted deep to the medial canthus of the eye towards the medial bony orbit (Braun and Casteel, 1993). The *femoral vein* may be accessed for phlebotomy in anaesthetised animals and is located by palpating a pulse in the neighbouring femoral artery.

Intraperitoneal injections are administered similarly to other species. This route is advisable only in anaesthetised or severely debilitated patients due to the risk of visceral puncture if the pig moves unexpectedly. Fluids should be warmed before intraperitoneal administration due to the risks of iatrogenic hypothermia with injection of cool fluids.

Intraosseous access is possible in neonates. The technique is similar to other mammals.

Intubation

The peculiar anatomy of the porcine upper airways (see Fig. 8.5) makes intubation technically challenging. A relatively small endotracheal tube should be selected – a 6 mm tube for a 25 kg animal, 9 mm for a 50 kg animal, up to a 14–16 mm size for large boars or sows. Although a

larger tube would allow more gas flow to the patient, tracheal damage from a large tube may cause haemorrhage or inflammation that will restrict ventilation during anaesthesia and the recovery period. The short porcine neck necessitates a short endotracheal tube, to avoid blockage of the epiarterial bronchial root and poor ventilation to the right apical lung lobe (Hodgkinson, 2007).

After induction of anaesthesia, the mouth is opened by pulling the upper and lower jaws apart with bandage material (Fig. 8.8). The head is held with the neck extended. The tongue is pulled forward, taking care not to cause iatrogenic damage on the sharp teeth. It can be difficult to visualise the larynx in pigs and it may help to have the animal in dorsal recumbency, as pharyngeal tissue may fall away from the larynx under gravity (Hodgkinson, 2007). Some clinicians prefer the patient to be in sternal or lateral recumbency. A laryngoscope with size 1–4 straight Wisconsin blade is used to visualise the larynx. The soft palate often lies dorsal to the epiglottis and should be moved with the laryngoscope blade or a stylet before local anaesthetic (for example, 2% lidocaine [lignocaine]) is sprayed on to the larynx to avoid laryngeal spasm during intubation. Laryngospasm is common if anaesthetic depth is insufficient (Goldman et al., 2005).

A stylet or introducer (Portex Ltd, Hythe, Kent, UK; M&IE Dentsply, Exeter, UK) is used to stiffen the endotracheal tube and aid insertion into the trachea. In some cases it may be easier to insert the introducer into the trachea and use it as a guidewire to thread the endotracheal tube into the trachea. The endotracheal tube is advanced to the glottis with the tip angled towards the glottal opening. Due to the acute angle between oropharynx and trachea, it may be easier to pass the stylet into the larynx before passing the endotracheal tube over it into the trachea. With either approach, it is often necessary to rotate the endotracheal tube 180° as it passes into the larynx and resistance is encountered. After a further 1 to 2 cm advancement, rotate the tube back again to pass it down the trachea. The endotracheal tube should not be forced against resistance as haemorrhage, inflammation or laryngeal spasm may occur, sometimes fatally (Flecknell, 1996). If resistance is felt, withdraw the tube slightly and re-advance. The cuff is inflated once the tube is in position within the trachea.

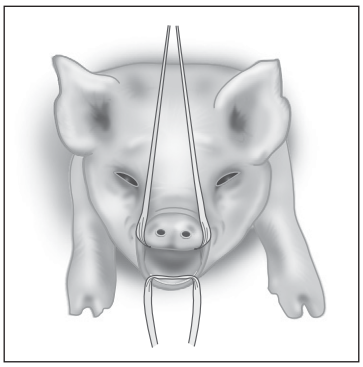


Figure 8.8 • Pig in sternal recumbency for intubation. Bandage material is used to open the jaws and extend the head to aid visualisation of the glottis.

As in rabbits, a laryngeal mask airway device (LMA-ProSeal®, Vitaid Ltd., Toronto, Canada) has been developed for use in pigs. This is simpler to place than an endotracheal tube, provides some protection for the pig's airway during anaesthesia and allows more reliable delivery of oxygen than a facemask or nasal catheter. However, unlike rabbits, pigs can vomit and the presence of some residual food, water or bedding material in the stomach means they cannot be considered starved. There is also a risk of gas leakage into the stomach with the laryngeal mask airway device, which is not found with endotracheal intubation. This may result in gastric distension and regurgitation. Regurgitation may lead to pulmonary aspiration, which is often fatal. The use of an oesophageal drainage tube to empty the stomach after induction and placement of the laryngeal mask airway device will reduce these risks (Goldman et al., 2005).

In unintubated animals, either a single or forked catheter, or pipe, can be inserted into the nasal cavity(ies) to provide oxygen and/or volatile anaesthetic agents. If endotracheal tubes are used, the cuffs can be inflated in the nasal passages. The mouth is taped closed to reduce oral breathing.

PRE-ANAESTHETICS

Sedatives are often administered prior to induction of anaesthesia to ease handling and reduce stress to the pig (Table 8.2). This greatly facilitates handling of the animals and enables intravenous induction techniques to be used. Pre-anaesthetic agents are usually administered intramuscularly, as intravenous injections are not possible in the fully conscious animal. If large volumes are involved, it may be useful to attach the syringe to a needle via an extension tube – this will reduce the restraint period to that required for initial insertion of the needle (Flecknell, 1996).

Anticholinergics, such as atropine and glycopyrrolate, are usually given to reduce salivary and bronchial secretions, abolish the vaso-vagal reflex during intubation and prevent bradycardia (Swindle, 1998).

Azaperone (Stresnil®, Janssen, High Wycombe, UK) is a butyrophenone derivative. It has minimal cardiovascular effects and is used specifically in pigs to cause sedation and ataxia. No analgesia is produced (Flecknell, 1996). The dose range is between 1 and 8 mg/kg, depending on the effect required; 1–2 mg/kg is usually administered intramuscularly to produce sedation for minor procedures. Azaperone pre-medication is administered 20–30 min before anaesthesia is induced by intravenous administration of another agent into the lateral auricular vein (Taylor, 2002). Azaperone at 5 mg/kg can be combined with metomidate at 2 mg/kg intramuscularly to produce deep sedation and analgesia for minor procedures.

During the onset period, the pig is left in a quiet area. Initial effects are observed in 5–10 min, peaking at around 20 min and lasting up to 1 h (Short, 1986; Swindle, 1998). Penile prolapse has been seen in boars given doses higher than 1 mg/kg (Hodgkinson, 2007). Azaperone causes peripheral vasodilation, which is useful for ensuing anaesthetic induction with intravenous agents, but will also increase heat loss. Insulation and supplemental heating should, therefore, be provided as appropriate.

Table 8.2: Sedatives and pre-medicants in pigs

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Alfaxalone/alphadalone	6[4]	IM	Sedation (use in small pigs)
Acepromazine	0.2–1.1[2]	IM	Tranquillisation
Atropine	0.04[3]	IM (or SC, IV)	Anticholinergic; 20–30 min prior to anaesthesia to reduce hypersalivation
Azaperone	0.25–0.50[2] 2.0[2] >2.0[8]	IM	Relaxation and mild sedation without ataxia Sedation and a degree of ataxia in 5–10 min, lasts 1 h Recumbency, possible adverse responses Doses <8 mg/kg reported for immobilisation[3,5]
Azaperone + metomidate	5 + 2[4]	IM	Deep sedation with analgesia for minor procedures
Diazepam	0.5–8.5[9]	IM, IV	Sedation; tranquillisation for minor procedure
Fentanyl/droperidol (Innovar-Vet®, Janssen)	1 ml/9–25 kg[2,10]	IM	Sedation; maximum effect in 20 min
Glycopyrrolate	0.005–0.010[3]	SC, IM, IV	Anticholinergic
Ketamine + xylazine	1−2+0.5–2.0[6]	IV	Tranquillisation/sedation. (Sufficient for Caesarean section when used with local anaesthetic at incision)
Midazolam	0.1–2.0[4,11]	IM	Sedation
Tiletamine/zolazepam	4–6[3,5]	IM	Sedation; immobilisation Doses <20 mg/kg reported[4]
Tiletamine/zolazepam/ ketamine/xylazine*	0.006–0.13 ml/kg[7]	IM	Tranquillisation/sedation
Xylazine	0.5–3.0[1]	IM	Sedation; tranquillisation

Key: IM = intramuscular, IV = intravenous, SC = subcutaneous

* Reconstitute 500 mg tiletamine/zolazepam powder (Telazol®, Fort Dodge Laboratories Inc, Fort Dodge, IA) with 2.5 ml of 100 mg/ml ketamine and 2.5 ml of 100 mg/ml xylazine

[1] (Braun, 1995); [2] (Braun and Casteel, 1993); [3] (Calle and Morris, 1999); [4] (Flecknell, 1996); [5] (Heard, 1993); [6] (Johnson, 1993); [7] (Ko et al., 1993); [8] (Short, 1986); [9] (Tynes, 1998); [10] (Wertz and Wagner, 1995); [11] (Swindle, 1993)

Ketamine at 10mg/kg intramuscularly will produce immobilisation, but spontaneous movements occur and it is more frequently used in combination with other agents. Higher doses are required in young animals (20 mg/kg) (Flecknell, 1996). Ketamine may be administered intramuscularly with azaperone, with or without butorphanol, to produce deeper sedation than azaperone alone (Hodgkinson, 2007).

The *phenothiazine* acepromazine is usually administered intramuscularly, as intravenous injection may lead to venous thrombosis (Hodgkinson, 2007). The sedative effects of acepromazine persist for 8–24 h (Hedenqvist and Hellebrekers, 2003). Sedative effects are unpredictable and side effects are similar with azaperone. Acepromazine may also be used in combination with ketamine.

Benzodiazepines, such as diazepam or midazolam, can be administered intramuscularly or intravenously, but intramuscular bioavailability of diazepam is poor. These drugs produce good muscle relaxation. Sedation is unreliable

when used alone, but is improved when administered with ketamine (Hodgkinson, 2007).

Midazolam or ketamine is often used to produce sedation, allowing intravenous catheter placement for induction with another injectable agent (Hedenqvist and Hellebrekers, 2003). Diazepam or midazolam (1–2 mg/kg intramuscularly) will produce rapid sedation and can be followed by ketamine to produce immobilisation (10–15 mg/kg intramuscularly) (Flecknell, 1996). *Tiletamine and zolazepam* (Zoletil®, Virbac, Carros, France; Telazol®, Fort Dodge Animal Health, IA, USA) is more useful in larger animals due to the small volume required. High doses (20 mg/kg) produce heavy sedation and immobilisation.

The *alpha-2-agonist* xylazine may be used as a sole agent to produce sedation and some analgesia. Both medetomidine and xylazine produce variable sedation and are more useful in potentiation of other agents (Flecknell, 1996). They may be used in combination with ketamine, ketamine and butorphanol, or tiletamine/zolazepam (Braun and Casteel, 1993). Alpha-2-agonists may induce vomiting.

Alphaxalone/alphadalone produces sedation in small pigs at 6 mg/kg intramuscularly (Flecknell, 1996). (It is not useful in larger animals due to the large volume of injection, for example 10 ml for a 20 kg pig.)

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Injectable anaesthetics

If anaesthesia is to be induced with injectable agents, it is common to pre-medicate the pig as described in the section above. This will allow intravenous injection to produce anaesthesia, usually into the auricular vein. Where a pre-medication has been given, the dose of anaesthesia for induction is reduced by 30–50% (Flecknell, 1996).

Ketamine (manufactured as a sole agent) and tiletamine (manufactured in combination with zolazepam) are *dissociative anaesthetic* agents commonly used in pig anaesthesia. Neither results in much muscle relaxation and if used alone will lead to rough recoveries from anaesthesia. For this reason, they are commonly used in conjunction with other agents.

The advantages of ketamine are good analgesia, a wide safety margin and the ability to administer the drug via the intramuscular or intravenous routes. Disadvantages include mild cardiovascular depression and muscle rigidity. When used alone, excessive salivation may also occur and recoveries are rough. Ketamine alone is insufficient for surgical anaesthesia (Boschert et al., 1994), but can be used in combination with agents such as benzodiazepines, azaperone or alpha-2-agonists to improve the anaesthesia produced. The addition of a benzodiazepine will produce muscle relaxation and good visceral analgesia, but hypothermia is profound (Hedenqvist and Hellebrekers, 2003).

Anaesthesia can be induced with intravenous ketamine at 2–5 mg/kg after pre-medication with azaperone (Taylor, 2002). After a loading dose of ketamine or other agents, ketamine is also commonly used to top up sedative or anaesthetic regimes, given at 5 mg/kg boluses or a continuous infusion (Hodgkinson, 2007).

Tiletamine and zolazepam will produce anaesthesia for minor surgery lasting 20 min (Hedenqvist and Hellebrekers, 2003). Used alone, this preparation causes poor muscle relaxation and recoveries may be rough. The zolazepam in the mix may prolong recovery. The addition of other agents produces a more balanced anaesthesia, with smoother induction and recovery (Table 8.3). To increase the proportion of dissociative agent, ketamine and xylazine rather than sterile water may be added to the powder for reconstitution (Thurmon and Benson, 1993).

Propofol is administered intravenously at 2.5–8 mg/kg to produce surgical anaesthesia lasting 10 min (Flecknell, 1996; Hodgkinson, 2007). Slow administration to effect should avoid the common dose-related adverse reaction of apnoea. This agent produces no analgesia, has a narrow safety margin, will reduce myocardial contractility and produces severe hypotension in pigs (Hedenqvist and Hellebrekers, 2003). Anaesthesia is commonly maintained via intubation and the use of volatile agents, but incremental injections or continuous rate infusion of propofol may be used. Recovery is smooth and rapid.

A lower dose of propofol may be administered with alfentanil. This provides analgesia, but respiratory depression will necessitate assisted ventilation. The alfentanil can be reversed with the opioid agonist-antagonist nalbuphine, administered intravenously at 0.5 mg/kg (Flecknell, 1996).

Barbiturates may be used to prolong anaesthesia after ketamine induction (Hedenqvist and Hellebrekers, 2003). They will inhibit respiration and ventilatory support may be required if barbiturates are used for anaesthesia.

Thiopental can be administered intravenously after sedation. The dose varies between animals and level of sedation, and is given to effect (Hodgkinson, 2007). This agent causes the least cardiovascular depression of the barbiturates (Hedenqvist and Hellebrekers, 2003). Surgical anaesthesia lasts 5–10 min (Flecknell, 1996). Unless a sedative or tranquilliser has been used, recovery may involve excitement in some patients.

Pentobarbital has been used in emergencies, but overdoses will result in severe cardio-respiratory depression. The safety margin appears to be higher in small pigs. Anaesthesia has been induced with intravenous pentobarbital after pre-medication with azaperone (Hodgkinson, 2007; Taylor, 2002). Administration should be slow, as this agent crosses the blood–brain barrier more slowly than thiopental. Surgical anaesthesia is provided for 10–30 min and top-up doses can be given, but prolong recovery. Full recovery may take up to 3 or 4 h (Flecknell, 1996).

Alphadolone/alphoxolone are *steroid anaesthetics* that can be administered intramuscularly or intravenously (1–2 mg/kg), usually after azaperone pre-medication. This produces anaesthesia lasting approximately 15 min,

Table 8.3: Anaesthetic agents in pigs

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Detomidine + butorphanol + midazolam + atropine	0.125 + 0.3 + 0.3 + 0.06[2]	IM	Anaesthesia. (Can reverse detomidine with yohimbine or atipamezole, butorphanol with naloxone, and midazolam with flumazenil)
Diazepam + ketamine	1–2 + 12–20[1]	IM	As for xylazine + ketamine dose below; longer anaesthesia, but diazepam not analgesic
Fentanyl + isoflurane	0.03–0.1mg/kg/h infusion and 0.050 mg/kg bolus + 0.5 %[9]	IV, Inhal	Anaesthesia using combination of opioid CRI and volatile agent
Guaifenesin + ketamine + xylazine*	0.5–1.0 ml/kg[6] 2.2 ml/kg/h[6]	IV	Administer to effect for induction Maintenance
Halothane	 4–5%[1] 1.5–2.5%[7]	Inhal	May cause malignant hyperthermia in some animals (genetic predisposition) Mask induction Maintenance
Isoflurane	4–5%[1] 2–3 %[7]	Inhal	Mask induction Anaesthesia maintenance
Ketamine	1–4 (prn)[1,5]	IV	Used to prolong anaesthesia after induction with ketamine combinations (see below)
Ketamine + acepromazine	10–20 + 0.05–0.50[2]	IM	Anaesthesia
Ketamine + diazepam	10–20 + 1–2[1,11]	IM	Administer diazepam first, giving ketamine when signs of sedation are seen Anaesthesia is short-term Smoother recovery than ketamine alone
Ketamine + xylazine	12–20 + 2.2[1]	IM	Administer xylazine first Xylazine enhances muscle relaxation and analgesia Short-term anaesthesia
Ketamine + xylazine + butorphanol	11 + 2 + 0.22[2]	IM	Anaesthesia Butorphanol enhances analgesia
Neostigmine	40–60 μg/kg[8]	IV	Reversal of non-depolarising neuromuscular blocking agents. (Dog/cat dose)
Nitrous oxide	50% inspired gases (with oxygen)[10]	Inhal	Calms patient for mask induction Also reduces concentration of other volatile agent required
Pancuronium	0.06[3]	IV	Neuromuscular blocking agent Reverse with neostigmine

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Pentobarbital	5–15[5]	IV	Anaesthesia induction after pre-medication Slow administration Top-ups prolong recovery
Propofol	2.5–8[3,5]	IV	Anaesthesia induction, usually after pre-medicant sedation (e.g. ketamine 10 mg/kg IM). Apnoea common, intubation advised
	8–12mg/kg/h[3,4]		Maintenance by CRI
Propofol + alfentanil[3]	2.0–2.5mg/kg + 20–230 μg/kg	IV	Induction (after ketamine 10 mg/kg pre-medication)
	0.1–0.2 mg/kg/min + 25 μg/kg/min		Maintenance
Sevoflurane	Prn[9]	Inhal	Mask induction/maintenance
Sufentanil + isoflurane	0.015–0.030mg/kg/h infusion and 0.007 mg/kg bolus + 0.5%[9]	IV + inhal	Anaesthesia using combination of opioid CRI and volatile agent
Thiopental	5–15[5]	IV	Anaesthesia induction after pre-medication Irritant extravascularly
Tiletamine/zolazepam/ ketamine/xylazine**	0.020–0.026ml/kg[7]	IM	Dissociative anaesthetic Short induction time, allows intubation
	0.022–0.044ml/kg1		Maintenance
Tiletamine/zolazepam + xylazine			Rapid induction, poor muscle relaxation, may have rough recovery
	2 + 2[1]	IV	Anaesthesia 30–40 min
	2–6 + 2.2[1,2,6]	IM	Can administer xylazine first, followed by tiletamine/ zolazepam when signs of sedation seen
Vecuronium	0.15[3]	IV	Neuromuscular blocking agent Reverse with neostigmine
Yohimbine	0.125–0.3[1,2]	IV	Reversal of xylazine and detomidine

Key: CRI = continuous rate infusion, Inhal = inhalation, IM = intramuscular, IV = intravenous, prn = dose to effect

*5% guaifenesin with 1–2 mg/ml ketamine and 1 mg/ml xylazine

** Reconstitute 500 mg tiletamine/zolazepam powder (Telazol®, Fort Dodge Laboratories Inc, Fort Dodge, IA) with 2.5 ml of 100 mg/ml ketamine and 2.5 ml of 100 mg/ml xylazine

[1] (Braun and Casteel, 1993); [2] (Calle and Morris, 1999); [3] (Flecknell, 1996); [4] (Goldman et al., 2005); [5] (Hodgkinson, 2007); [6] (Johnson, 1993); [7] (Ko et al., 1993); [8] (Lukasik, 1995); [9] (Swindle, 1998); [10] (Tynes, 1998); [11] (Wertz and Wagner, 1995)

although a continuous infusion can be used to prolong anaesthesia. Cardiovascular depression may be seen, but respiratory depression is minimal and muscle relaxation is good (Hodgkinson, 2007).

Opioids, such as fentanyl or sufentanil, can be administered via continuous rate infusion in combination with inhalant agents to maintain anaesthesia. Opioid infusions do not decrease myocardial contractility or coronary blood flow (Hedenqvist and Hellebrekers, 2003).

Although not routinely used, the *neuromuscular blockers* pancuronium and vecuronium can be administered to produce muscular paralysis in pigs for certain procedures. Inadequate anaesthesia is assessed by changes in heart rate and blood pressure (Hedenqvist and Hellebrekers, 2003).

Volatile anaesthetics

Inhalant anaesthetic agents are considered safer than injectable anaesthetics in pigs. Volatile agents appear to be less potent in pigs and comparatively higher concentrations are required than in other species (Flecknell, 1996). Isoflurane and halothane have wide safety margins, short induction and recovery times, and the depth of anaesthesia can be rapidly adjusted. Desflurane has also been used in pigs (Swindle, 1998). Halothane is associated with malignant hyperthermia in certain genetically predisposed commercial pig breeds (Claxton-Gill et al., 1993). Isoflurane has the least myocardial depressant effects of the volatile anaesthetic agents (Hedenqvist and Hellebrekers, 2003).

Small pigs may be induced using a facemask and volatile agents, but larger animals require pre-medication before they can be restrained for application of a mask. As in other species, volatile agents may be administered via a closely fitting facemask or endotracheal tube. It can be difficult to create a tight seal and environmental contamination with anaesthetic gases is likely to occur. After induction it is, therefore, preferable to intubate the patient in order to ensure airway patency and allow positive pressure ventilation (PPV) to be performed (Swindle, 1998). This is particularly useful if laryngospasm or respiratory obstruction occurs. However, intubation can be technically difficult and facemasks are suitable for short procedures. Equipment (local anaesthetic, laryngoscope and endotracheal tubes) should always be ready in case it becomes necessary to intubate the patient.

Nitrous oxide may be added to the inhaled gases, up to 50%, to reduce the concentration of other agents required (Hedenqvist and Hellebrekers, 2003). Administration of nitrous oxide with oxygen (50:50) may be beneficial in calming the patient during mask induction before the addition of other volatile agents (Tynes, 1998). *Halothane* can be used to induce and maintain anaesthesia in pigs. *Isoflurane* results in more rapid induction and recovery of anaesthesia, but has a more pungent odour than halothane and may be resented if used for induction by facemask (Hodgkinson, 2007). Adult pigs usually require an oxygen flow rate of 1–3 L/min and can be maintained on 1.5–2.5% halothane or 2–3% isoflurane.

Anaesthetic maintenance

Volatile agents are usually used for maintenance of anaesthesia, via an endotracheal tube or close-fitting facemask. Alternatively, injectable agents may be used, for example a continuous rate infusion of propofol (8–12 mg/kg/h) and hourly boluses of ketamine (1–4 mg/kg) (Braun and Casteel, 1993; Flecknell, 1996; Goldman et al., 2005).

Recovery

As pigs are prone to laryngospasm, they should be closely monitored during recovery until able to sit in sternal recumbency.

Group animals should be kept separate from companions during the recovery phase to avoid trauma from conspecifics (Hodgkinson, 2007). If this is not possible, the whole group should receive azaperone to prevent fighting (Taylor, 2002).

Suggested anaesthetic protocols

In general, injectable agents are used to induce or assist in induction of anaesthesia before maintenance with volatile anaesthetic agents. If endotracheal intubation or venous catheterisation cannot be attained, anaesthesia is maintained via further intramuscular agents or volatile agents via a facemask (Hodgkinson, 2007).

Pre-medication with atropine (0.04 mg/kg), azaperone (4 mg/kg), midazolam (0.2 mg/kg) and ketamine (2 mg/kg) is administered intramuscularly. At the same time, local anaesthetic cream is applied to the ear to allow placement of an intravenous catheter in the auricular vein 20–30 min later. Anaesthesia is induced with intravenous propofol at 2–3 mg/kg (ketamine combinations are a common alternative), using a butterfly catheter with extension tubing in the auricular vein (Taylor, 2002). Since apnoea is common after propofol, the patient should be intubated and PPV performed if necessary. Anaesthesia is maintained with isoflurane. If volatile anaesthesia is not available, a continuous rate infusion of propofol (0.2 mg/kg/min) and hourly boluses of ketamine (1–2 mg/kg) can also be used to maintain anaesthesia (Goldman et al., 2005).

Ideally, the pig should be intubated after induction. If intubation is not possible, oxygen and volatile anaesthetic agents can be provided to the pig via a facemask over the snout, or via a single or double nasal catheter. Nitrous oxide (2:1 ratio of oxygen to nitrous oxide) can be used during maintenance of anaesthesia to reduce the inspired concentration of volatile anaesthetic agent required. Denitrogenation should be performed by supplementing oxygen without nitrous oxide for 10 min at the end of anaesthesia.

ANAESTHESIA MONITORING

Observations on the patient

Positioning

As pig hearts are relatively small, with an associated small cardiovascular capacity, pigs should not be maintained in dorsal recumbency for long periods (Braun and Casteel, 1993).

Cardio-respiratory systems

The heart rate and pulse rate should be monitored and recorded throughout anaesthesia. Pulses may be difficult to palpate in larger thick-skinned animals, but can usually be detected at several arteries, including the auricular, carotid, femoral, saphenous or (in anaesthetised animals) sublingual arteries. The carotid and femoral arteries are deep in muscle and only palpable in small pigs (Braun and Casteel, 1993).

Capillary refill time can be monitored to assess the peripheral circulation. Mucous membrane colour will give the anaesthetist some appreciation of oxygen saturation. A pulse oximeter can be applied to the ear, tongue or tail to assess oxygen saturation of haemoglobin during anaesthesia.

The respiratory rate should be monitored by observation of thoracic excursions or movement of the reservoir bag if the patient is intubated.

Pigs are prone to laryngospasm and should be closely monitored when unintubated (during anaesthesia or recovery). Steroids should be administered and intubation or tracheostomy performed if spasm occurs.

Nervous system

Muscle tone, including jaw tone, is a good indicator of anaesthetic depth. As with other mammalian species, ocular reflexes, such as corneal and palpebral, can be assessed. When under a light plane of anaesthesia, there will be a strong ear flick response to gentle stroking of hairs inside the pinna.

Anaesthetic monitoring equipment

Body temperature should be monitored with a digital rectal *thermometer*. Skin temperature should be assessed if malignant hyperthermia is considered possible, as this will result in hot, pink blotchy skin (Hodgkinson, 2007). In some cases, particularly small patients, supplemental heating will be required to prevent hypothermia. In others, body heat can be maintained by wrapping the body in bubble-wrap.

An *electrocardiogram (ECG)* can be used as in other species to record electrical activity from the heart. An *arterial catheter* in the superficial femoral artery will allow continuous invasive blood pressure monitoring and measurement of arterial blood gases (Goldman et al., 2005).

PERI-ANAESTHETIC SUPPORTIVE CARE, INCLUDING ANALGESIA

Although pigs have a layer of subcutaneous fat, *hypothermia* may be a problem during anaesthesia, even in large animals, due to their relative hairlessness. Performing the anaesthetic in a space without draughts, along with the use of blankets and bubble-wrap, should reduce heat loss. If the environmental temperature is cold, use methods of supplemental heating such as electric heat pads, warm air blankets and overhead heaters to increase the temperature. Young and small patients in particular should be closely monitored and extremities, such as limbs and ears, insulated (for example, with bubble-wrap) to prevent heat loss.

Malignant hyperthermia is a genetic-associated condition that has been reported in certain domestic pig breeds, including Landrace and Large White pigs, usually in response to halothane anaesthesia (Hodgkinson, 2007). The gene governing susceptibility can be tested for in a blood sample (Taylor, 2002). Other species are less likely to suffer from this condition, but core body temperature should be monitored during anaesthesia to ensure hyperthermia does not occur. If anaesthesia is performed outdoors, shade should be provided to protect the patient from sunburn. If hyperthermia is noted, cool fluids should be administered intravenously; colonic lavage can be helpful, as can ice packs, or wet towels can be applied to the skin.

Analgesia

Analgesia should be provided to animals with painful conditions or after surgery (Table 8.4). A combination of agents can be used (multimodal analgesia). For example, the use of local anaesthesia along an incision line with parenteral opioid analgesics and anaesthesia results in a reduction in the effects of noxious stimuli as well as speeding recovery times from painful procedures (Swindle, 1998).

Local anaesthetics

These drugs are useful for several techniques in pigs. As described above, *topical* application of lidocaine (lignocaine) is used to reduce the risk of laryngospasm during endotracheal intubation. Local anaesthetics, such as 5% procaine, can be *infiltrated locally* for minor procedures or used in combination with sedatives for a more involved procedure. For anaesthesia of the testicles prior to castration of pigs up to 6 months old, 2–15 ml of 5% procaine can be injected subcutaneously and intratesticularly. The patient should be left for at least 5 min to allow the anaesthetic effects to occur before surgery is initiated (Hodgkinson, 2007).

Intravenous regional anaesthesia can be performed as in other animals. A tourniquet is applied around the proximal limb to slow systemic spread and local anaesthesia injected into a superficial vein. This can be useful for distal limb surgery. Manual restraint with or without sedation will also be required (Hodgkinson, 2007).

Table 8.4: Analgesics in pigs

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)/COMMENT
Local anaesthetics			
Lidocaine (lignocaine) 2%	–	Topical	Spray on to larynx 2 min before intubation to avoid laryngospasm
Lidocaine/prilocaine (EMLA® cream, AstraZeneca)	–	Topical	Apply to auricular skin at time of pre-medication, to ease intravenous access for induction Allow 20–30 min to take effect
Prilocaine 5%	2–15 ml[3]	SC or into tissue	–
Non-steroidal anti-inflammatory drugs			***Anti-inflammatory, analgesic, antipyretic*** ***Many side effects (see text)***
Carprofen	2–4[1]	IV, SC	24
Flunixin	0.5–1.0[2]	SC, IV	12–24
Ketoprofen	3[3]	IM	–
Meloxicam	0.4[3]	IM	–
Phenylbutazone	4–8[a]	PO	12;
Opioids			
Buprenorphine	0.005–0.010[4]	IM, IV	8–12
Butorphanol	0.1–0.3[4]	IM, IV, SC	8–12
Fentanyl	0.02–0.05[5]	IM, IV	–
Morphine	0.2[2]	IM, SC	4
Pentazocine	2–5[5]	IM	4
Pethidine (meperidine)	2–10[2]	IM	4

Key: IM = intramuscular, IV = intravenous, PO = oral, SC = subcutaneous

[1] (Flecknell, 1996); [2] (Heard, 1993); [3] (Hodgkinson, 2007); [4] (Swindle, 1993); [5] (Swindle and Adams, 1988)

Lumbosacral extradural anaesthesia can be performed in pigs similarly to ovines, and is useful for obstetrical and perineal procedures. The bony landmarks of wings of the ileum are more difficult to palpate in larger animals and sedation is advisable. A 19-gauge, 13 mm needle is necessary in an adult pig; 2% lidocaine (lignocaine) is administered into the extradural space, at 1 ml per 7.5 kg up to 50 kg, then 1 ml per 10 kg body weight above this. This dose will result in recumbency for up to 2 h (Hodgkinson, 2007).

Non-steroidal anti-inflammatory drugs

Pigs are susceptible to the side effects of non-steroidal anti-inflammatory drugs (NSAIDs) and repeated administration may not be advisable. NSAIDs are contraindicated in patients with renal, hepatic or cardiac disease. Gastrointestinal ulceration may also be seen. Therefore, it is advisable to administer a gastrointestinal protectant simultaneously, for example ranitidine (150 mg per animal orally every 12 h) (Baldrick, 1993), particularly if more than one dose of NSAID is to be given.

Carprofen has been used to control pain post-operatively in pigs for up to 3 days without side effects (Dobromylskyj et al., 2000). Aspirin has been used orally in pigs (Hedenqvist and Hellebrekers, 2003).

Opioids

The analgesic effects of buprenorphine have been demonstrated in pigs (Dobromylskyj et al., 2000). Both buprenorphine and butorphanol can be used in pigs with few side effects (Hedenqvist and Hellebrekers, 2003).

DRUG DOSES, ROUTES OF ADMINISTRATION, DURATION OF ACTION

Drugs licensed for pigs will vary between countries. Legislation is usually less stringent for pet animals than those that will enter the food chain. There may be breed

Table 8.5: Emergency drugs in pigs

DRUG	DOSE (mg/kg)	ROUTE	INDICATION/ COMMENT
Atropine	0.04[1]	SC, IM, IV	Bradycardia; hypersalivation
Dantrolene sodium	2–5[2]	PO, IV	Malignant sodium hyperthermia Repeat q8h until normothermic
Dexamethasone	0.1[3]	IM, IV	Shock; laryngeal oedema
Glycopyrrolate	0.005–0.010[1]	SC, IM, IV	Bradycardia; hypersalivation
Naloxone	4 mg total dose[1]	IV	Opioid reversal

Key: IM = intramuscular, IV = intravenous, PO = oral, q8h = every 8 hours, SC = subcutaneous

[1] (Calle and Morris, 1999); [2] (Claxton-Gill et al., 1993); [3] (Murison, 2001)

and individual animal differences between drug doses. Agents that can be administered to effect, such as agents administered by inhalation or intravenous injection, are thus often used. This is particularly important when anaesthetising a patient with unknown health status.

EMERGENCY PROCEDURES/ DRUGS

Respiratory problems

Brachycephalic breeds, such as Vietnamese Potbellied pigs, are prone to respiratory obstruction (Hodgkinson, 2007). This can occur during sedation or in unintubated patients. Appropriate sizes of endotracheal tube and a long-bladed laryngoscope should, therefore, be ready to achieve airway patency if required. If anaesthesia is being performed in the veterinary practice, oxygen can be administered from an anaesthetic machine. If the anaesthetic is outwith the practice premises, self-inflating bag-valve-masks (Ambu-bag) or resuscitators can be used to provide PPV with environmental air in such cases. Placing the pig in dorsal recumbency may assist with alleviating the obstruction (as with intubation).

Laryngospasm may also occur during recovery. If dyspnoea or restlessness is noted, intubate the animal or perform a tracheotomy. These procedures may require re-anaesthetisation (Hodgkinson, 2007). Steroids should be administered at anti-inflammatory doses (Murison, 2001).

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9 Avian anaesthesia

INTRODUCTION

There are over 9800 species of bird within 28 orders (Perrins, 2004), and many species are kept as pets. This chapter will discuss the avian anatomy and physiology pertinent to anaesthesia of these species, along with anaesthetic drugs and techniques in birds. Subsections will provide more details on groups commonly kept as pets, with birds of prey (Falconiformes and Strigiformes) discussed separately from other orders (Passeriformes, Psittaciformes and Columbiformes). There is much anatomical and physiological variation even within one order, with, for example, nearly 6000 species of Passeriformes (King and McLelland, 1984).

Manual restraint often causes stress that may be fatal to the avian patient and anaesthesia is frequently used as a less stressful alternative in many procedures for birds. Non-painful procedures, such as venepuncture, radiography and ophthalmoscopy, require a more simplistic approach than painful procedures, such as surgery. The anaesthetic requirements vary depending on the procedure to be performed and the species involved. Many anaesthetic and analgesic studies have been performed in various bird species, but the existence of species differences in response to drugs may reduce the application of this work to other species and increases the requirement for further research. Although broad anaesthetic principles can be applied across many avian species, drug doses may vary significantly between species. The reader is referred to other texts for additional references regarding individual species.

Certain aspects of avian anatomy and physiology, particularly the cardiovascular and respiratory systems, are markedly different to mammal systems and are associated with increased anaesthetic risks in birds. Although volatile anaesthetic agents and improved monitoring techniques have significantly reduced the risks associated with anaesthetising birds, the procedure is never risk-free.

If a prolonged anaesthesia is anticipated or becomes necessary, the side effects of anaesthesia will affect the patient more than during a short procedure. During longer anaesthetics, closer attention to stabilising the patient and maintaining physiological parameters is necessary to avoid peri-anaesthetic mortalities. As with other exotic pet species, pre-anaesthetic assessment and peri-anaesthetic care are of paramount importance in reducing morbidity and mortality. It is often necessary to stabilise birds for up to 48 h before anaesthesia is induced. Supportive care and protocols for anaesthesia in various bird species presented to veterinary practices will be described.

The aim of this chapter is to provide the veterinary surgeon with knowledge of avian anatomy, physiology and common conditions that may affect risks associated with anaesthesia of the patient. It will also describe anaesthetic-related techniques and drugs for birds. Later chapters identify differences between avian species.

ANATOMY AND PHYSIOLOGY

As the basic anatomy and physiology of birds are very different from mammals, this section will describe the main differences. There are many variations between bird species.

Temperature

Birds are endothermic, but their core body temperature is much higher than that of mammals, between 39°C and 42°C (Dawson and Whittow, 2000). Allied to this high temperature is a high metabolic rate, which provides sufficient energy to permit flight (Dorrestein, 1997). Birds with insufficient energy resources to maintain the high metabolic rate will have difficulty in maintaining their body temperature. Therefore, ill birds should be hospitalised in a warm environment or incubator to reduce their energy requirements.

Avian species also have a low tolerance for high temperatures and die at 46°C (O'Malley, 2005). Birds use

Table 9.1: Common pet avian species

SPECIES	ORDER	AVERAGE WEIGHT
African grey parrot (*Psittacus erithacus*)	Psittaciformes	310–460 g
Barn owl (*Tyto alba*)	Strigiformes	262–600 g
Blue and gold macaw (*Ara ararauna*)	Psittaciformes	850–2000 g
Blue-fronted Amazon (*Amazona aestiva*)	Psittaciformes	275–510 g
Budgerigar (*Melopsittacus undulates*)	Psittaciformes	35–85 g
Canary (*Serinus canaria*)	Passeriformes	12–29 g
Cockatiel (*Nymphicus hollandicus*)	Psittaciformes	70–108 g
Eagle owl (*Bubo bubo*)	Strigiformes	1.6–2.5 kg
Fisher's lovebird (*Agapornis fischeri*)	Psittaciformes	40–50 g
Goshawk (*Accipiter gentiles gentiles*)	Falconiformes	634–880 g (male); 980–1200 g (female)
Great Indian hill mynah (*Gracula religiosa intermedia*)	Passeriformes	180–260 g
Greater sulphur-crested cockatoo (*Cacatua galerita galerita*)	Psittaciformes	500–1250 g
Hahn's macaw (*Ara nobilis*)	Psittaciformes	150–180 g
Harris hawk (*Parabuteo unicinctus*)	Falconiformes	574–1000 g
Kestrel (*Falco tinnunculus*)	Falconiformes	145–167 g (male); 193–282 g (female)
Lanner falcon (*Falco biarmicus*)	Falconiformes	500–600 g (male); 700–900 g (female)
Lesser sulphur-crested cockatoo (*Cacatua sulphurea*)	Psittaciformes	228–315 g
Rainbow lorikeet (*Trichoglossus haematodus*)	Psittaciformes	120–130 g
Merlin (*Falco columbarius*)	Falconiformes	160–170 g (male); 220–250 g (female)
Racing pigeon (*Columba livia*)	Columbiformes	230–540 g
Saker falcon (*Falco cherrug*)	Falconiformes	680–990 g (male); 970–1300 g (female)
Scarlet macaw (*Ara macao*)	Psittaciformes	1–1.5 kg
Senegal parrot (*Poicephalus senegalus*)	Psittaciformes	125–150 g
Sparrow hawk (*Accipiter nisus*)	Falconiformes	150–210 g (male); 190–300 g (female)
Yellow rosella parakeet (*Platycercus flaveolus*)	Psittaciformes	100–120 g
Zebra finch (*Poephila guttata*)	Passeriformes	10–16 g

(Coles, 1997; O'Malley, 2005)

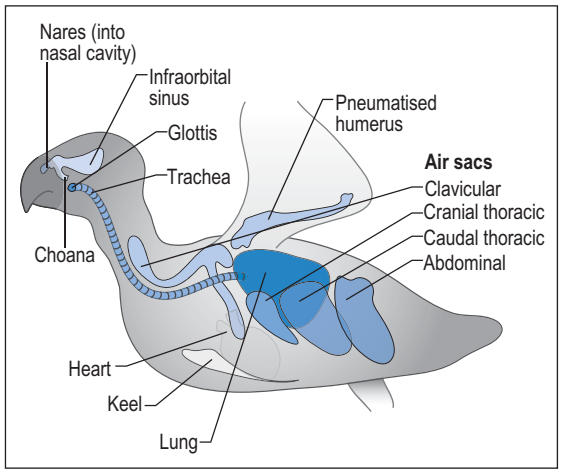


Figure 9.1 • Respiratory system in a parrot (after Harcourt-Brown, 2005)

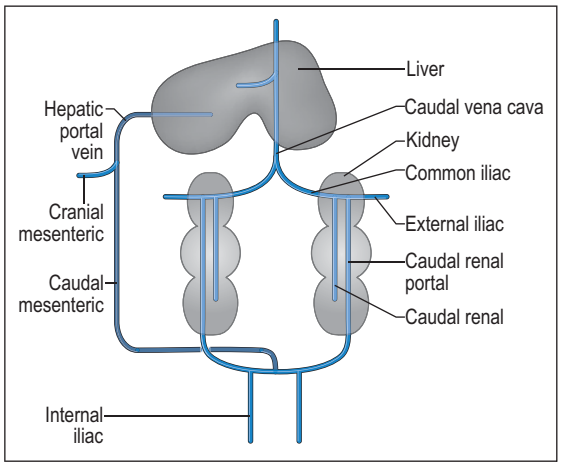


Figure 9.2 • Major caudal veins in the bird, including the renal portal system.

several techniques to lose heat: thermal panting and gular fluttering, via the large surface area of the air sacs, via convection from thinly feathered skin on the ventral wings, through the skin, or via blood shunts (Dawson and Whittow, 2000; Welty, 1982).

As birds use their plumage in thermoregulation, any problems with their feathers, for example oiling or loss due to feather plucking, will result in a reduced capacity to regulate their body temperature. Birds such as this require extra care during hospitalisation to ensure they do not succumb to hypothermia.

Cardiovascular system

The four-chambered heart lies slightly to the right in the midline, cranially within the thoracoabdominal cavity close to the sternum (Fig. 9.1) (Akester, 1984). In some species the ascending aorta curves to the right or the vena cavae enter the right atrium via a sinus venosus (King and McLelland, 1984; Rosenthal et al., 1997). There are also minor differences in the valves and cardiac electrical conduction system in birds compared to other animals. Type 2 Purkinje fibres completely penetrate the ventricular myocardium, assisting synchronous beating at high rates (Edling, 2006).

Avian hearts are considerably larger than similar-sized mammals, due to their high oxygen demands (Maina, 1996). The pericardium normally contains a small amount of fluid and attaches to the sternum dorsally (Pees et al., 2006). The lungs are dorsal to the heart (Smith and Smith, 1997). As birds have no diaphragm, the liver lobes surround the apex of the heart.

The higher cardiac output in birds is produced by a combination of factors, including a high stroke volume, fast heart rate (typically 150–350 beats per minute at rest), and slightly lower peripheral resistance (Sturkie, 1986). Arteries are stiffer than in mammals, maintaining high blood pressure and improving blood flow. A consequence of this high pressure is an increased risk of fatality due to aortic rupture, heart failure and haemorrhage in stressed patients (Rosenthal et al., 1997; Welty, 1982).

The heart is innervated by sympathetic and parasympathetic nerves. The main sympathetic neurotransmitters adrenaline (epinephrine) and noradrenaline (norepinephrine) (Sturkie, 1986) are increased with excitement. This may be significant, for example, with volatile anaesthetic agents that may sensitise the myocardium. Cardiovascular function is depressed by hypoxia, hypercapnia and anaesthetics (Edling, 2006).

Birds have a renal portal system similar to reptiles (Fig. 9.2) (O'Malley, 2005). Venous blood drains from the legs, caudal body and gastrointestinal tract into the caudal vena cava via this system (Smith and Smith, 1997). Valves and the presence of smooth muscles in the blood vessels allow blood to pass through the kidneys when the valves close and to bypass the kidneys at times of stress when adrenaline (epinephrine) release opens the valves (Akester, 1971; West et al., 1981). The large coccygeal mesenteric vein also drains the hindgut mesentery, from the hepatic portal vein to the renal portal vein; blood flow in this vein can be bi-directional, allowing blood to flow between the kidneys and liver. Opening of the portal valves allows blood to follow the latter route to the liver or directly into the caudal vena cava to the heart.

The total blood volume varies between 5% and 13% of body mass in birds (O'Malley, 2005). Although birds rapidly regenerate erythrocytes, blood samples should not be greater than 8% of the total circulation in healthy birds (less in ill animals). As conscious restraint is very stressful for most birds and veins are fragile, venepuncture is most commonly performed under anaesthesia. The spleen is not used to store blood in birds, and splenic contraction does not occur after blood loss (Coles, 1997).

The right jugular vein (see Fig. 9.4) is usually larger than the left, as an anastomosis at the angle of the jaw slopes

caudally to the right (Akester, 1971; West et al., 1981). This vein is commonly used for phlebotomy or administration of fluid boluses in small to medium-sized birds. Most birds have a featherless tract, or aptera, over this vein; Columbiformes and Anseriformes (mainly waterfowl) lack these apteria (Echols, 1999).

Cardiovascular disease has been reported in many avian species, but is mostly of case reports rather than large studies. These include congenital defects, pericardial diseases, myocardial or endocardial disease, arteriosclerosis in psittacines (Fricke et al., 2003; Hasholt, 1969) and circulatory collapse. Nutritional imbalances may be associated with cardiomyopathy in captive birds (Harrison and McDonald, 2006).

Clinical signs of cardiovascular disease may include dyspnoea, lethargy, weakness, exercise intolerance, syncope, abdominal distension or sudden death (Raftery, 2005). Cardiovascular assessment may be performed in birds, but normal reference values are limited. It is important to minimise handling and examination in these patients, as birds presenting with clinical signs usually have cardiac decompensation and stress may be fatal (Pees et al., 2006). Respiratory, renal and hepatic function may also be compromised secondary to a circulatory disorder. Radiography is used as a baseline to assess the cardiac silhouette, the large vessels and changes in other organs. Echocardiography is useful and may be possible in conscious patients (Krautwald-Junghanns et al., 1995; Straub et al., 2002). Electrocardiograms (ECGs) can also be used to assess electrical activity in the heart, but anaesthesia may be required. The reader is referred to other texts for therapeutic options of birds with cardiovascular disease.

Respiratory system

Breathing in birds is nasal or oral (Powell, 2000). The nares are lateral at the base of the beak, although the position and size vary between species, and some species have an operculum in each to prevent foreign body inhalation. (Some bird species, for example gannets [*Morus* sp.] do not have external nares, rather breathing through a narrow gap between the beaks (O'Malley, 2005).) Air enters the nasal cavity and thence via the choana to the oropharynx and the glottis (King and McLelland, 1984). The rete mirabile is a network of blood vessels within the nasal conchae, which assists with water and thermal homeostasis; nebulisation of avian patients can thus be used to aid rehydration. Sinusitis is common in pet birds and may be evidenced by swelling ventromedial to the orbit where the infraorbital sinus is superficial. This sinus communicates with the nasal conchae and the cervicocephalic air sac, which may allow dissemination of pathology.

The opening to the larynx is the glottis, which is attached midline to the base of the tongue. The exact positioning varies between species, but the glottis is usually easily visible on opening the oral cavity and intubation accomplished relatively easily. The lack of epiglottis in birds increases the risk of inhalation of foreign material if the trachea is not protected by intubation during anaesthesia.

Table 9.2: Average respiratory and heart rates for different sizes of bird

WEIGHT	RESPIRATORY RATE (breaths per minute)	HEART RATE (beats per minute)
40–100 g	55–75	600–750
100–200 g	30–40	450–600
250–400 g	15–35	300–500
0.5–1 kg	8–25	180–400
5–10 kg	2–20	60–70

(Adapted from Forbes and Altman, 1998)

Avian tracheal rings are complete and the tracheal lumen wide (1.3 times the tracheal width in a similar-sized mammal) (Coles, 1997). The trachea is relatively long, with an associated increased dead space volume compared to mammals (Hinds and Calder, 1971). To compensate for this, birds have an increased tidal volume (four times) and slower respiratory rate (one-third) compared to similarly sized mammals. For example, a 100 g bird will have a respiratory rate of approximately 30 breaths per minute, one-third the rate of a 100 g mammal (Welty, 1982). The syrinx is the sound-producing organ in birds and is sited at the tracheobronchial junction. Narrowing of the airway here predisposes this site for foreign body obstructions.

After the syrinx, the trachea bifurcates into the left and right primary bronchi, which run to the lungs and then intrapulmonic to the caudal air sacs. In most avian species, four sets of secondary bronchi arise from each bronchus within the lungs: these are the medioventral, mediodorsal, lateroventral and laterodorsal groups (McLelland, 1989). These secondary bronchi terminate in tertiary bronchi (parabronchi) (O'Malley, 2005). Smooth muscle, lining all bronchi, permits dilation and contraction (Lasiewski, 1972).

The major difference between the mammalian and avian respiratory system is the small lung size, with non-respiratory air sacs attached to aid airflow. Avian lungs are relatively rigid and quite dense, adhering to the dorsal rib cage. The lungs usually extend from the rib on the last vertebrae to the cranial edge of the ilium.

The avian lung is connected to the air sacs via the secondary bronchi at the ostium, which is usually ventrolateral (O'Malley, 2005). The air sacs are thin-walled and relatively avascular; they partake in less than 5% of gas exchange (Fedde, 1993; Magnusson et al., 1976). Most species have nine air sacs. These comprise paired cervical, a single clavicular (paired in some species), paired cranial thoracic, paired caudal thoracic and paired abdominal air sacs. The air sacs act as bellows, and produce unidirectional airflow through the lungs. The air sacs may be cannulated

(see Techniques section) and oxygen and/or anaesthetic gases administered into the respiratory system. The air sacs extend into the medullary cavity of some bones, including the humerus, sternum, coracoid, vertebrae and pelvis. In certain species, further pneumatisation may include the femur, scapula and furcula (Koch, 1973; Maina, 1996).

Birds do not have a diaphragm. However, a peritoneal sheet, the horizontal septum, lines the ventrum of the pleural cavities (Duncker, 1979). This septum does not assist with respiratory movements, but passively displaces the viscera. Respiratory movements are due to the external and internal intercostals and abdominal muscles, and both inspiration and expiration require active muscle movements (Ludders and Mathews, 1996; O'Malley, 2005). Contraction of the inspiratory muscles produces an increase in volume within the coelomic cavity; this causes pressure in the air sac to become negative relative to atmospheric pressure, and air flows into the respiratory system. Expiratory muscle contraction is the reverse, with air flowing back across the lungs and out of the bird's body. The system stops midpoint between inspiration and expiration during the resting phase (Edling, 2006). Due to the presence of air sacs, air will take two breaths to cycle through the respiratory tract (McLelland and Malony, 1983).

In one breath, 50% of inspired gases pass directly to the posterior air sacs, only passing through the lungs for gas exchange during the next breath (Fig. 9.3). The other 50% of inspired gases pass through the lungs and undergo exchange of gases during the first breath, but also accompany the gas from the posterior air sacs through the lungs for further gas exchange on the second breath. This mechanism increases the speed and efficiency of gas absorption into the blood. Volatile anaesthetic agents are thus absorbed rapidly in birds, with accompanying rapid changes in blood concentrations (Coles, 1997).

The central nervous system provides a rhythmic stimulus for respiration, modulated by various reflexes. Central and arterial chemoreceptors are sensitive to arterial carbon dioxide and pH changes; arterial chemoreceptors also respond to changes in arterial oxygen. Intrapulmonary chemoreceptors in birds are extremely sensitive to carbon dioxide and insensitive to hypoxia (Edling, 2006).

Gas exchange occurs in the avian parabronchi. They have invaginations (atria), leading to microscopic air capillaries (Powell and Scheid, 1989). Blood capillaries interlace the anastomosing network of air capillaries to enable gas exchange. Air flow may be either uni- or bidirectional depending, respectively, on whether the anatomy of the specific airway is ordered and parallel (in the paleopulmonic region) or irregularly branched (the neopulmonic) (Brackenbury, 1987; Powell, 2000; Scheid and Piiper, 1971). The ratios of paleopulmonic and neopulmonic sections vary between species. For example, the neopulmonic portion comprises 10–12% in pigeons, and 20–25% in passerines and psittacines (Edling, 2006; Ludders and Mathews, 1996).

Oxygen absorption in avian lungs is ten times more efficient compared to mammalian lungs, due to several avian adaptations. The avian air capillaries are much smaller than mammalian alveoli, with the diameter in avians less than one-third that in mammals (King and McLelland, 1984). There is a thin blood–gas barrier across the air capillaries in the lungs, and the rigid lungs allow 20% more gas exchange area compared to mammals (King and Molony, 1971; Scheid and Piiper, 1972). A cross-current exchange system is present, with blood flow at right angles to the unidirectional airflow, which allows continuous gas exchange to occur.

The normal avian Pa_{CO2} is lower than that in mammals due to the lung efficiency discussed in the last paragraph. Birds are very sensitive to hypercapnia. Gas flow rates during anaesthesia should, therefore, be higher for birds, at least three times the normal minute volume (Coles, 1997). Normal minute volumes for birds vary between species, but are approximately 800 ml/min/kg (Klide, 1973). As discussed in the introductory chapter, the flowmeter on most anaesthetic machines is inaccurate at low flow rates, so a minimum of 1 L/min should be used with small birds (increasing appropriately in larger patients).

Dorsal recumbency will allow compression of the caudal air sacs by viscera, reducing minute volume by up to 60%. This effect is less with birds in lateral recumbency (King and Payne, 1964). Positive pressure ventilation is recommended in anaesthetised birds, as they may not breathe regularly (Fedde, 1993).

Another important physiological difference is the small functional residual capacity (FRC) in birds compared to mammals. Although birds have a large respiratory gas volume for their body weight, only 10% of the total specific volume resides within the location of gas exchange (the parabronchi and air capillaries) at one time. With a small FRC, gas exchange will not occur without airflow. Periods of apnoea will rapidly upset the acid–base balance and become life-threatening (Edling, 2006).

Respiratory disease in birds is common. It may be due to a number of aetiologies, including bacterial, viral, fungal or yeast infection, hypothyroidism, airway obstruction by a foreign body or granuloma, or toxins. Malnutrition often predisposes respiratory disease by depressing the immune system and altering the function of epithelial surfaces, for example by causing loss of cilia and a decrease in mucus production (Harrison and McDonald, 2006). The airways may become obstructed by liths and secondary infections are common.

Clinical signs seen with respiratory disease may vary from a slightly quiet or 'fluffed-up' bird to tail-bobbing and open-mouth breathing. Signs associated with upper airway obstruction and requiring air sac cannulation include gasping for air and 'squeaks' produced with each breath. Birds with air sacculitis may have whole-body movement during respiration and crackles may be heard on lung auscultation (Harrison et al., 2006). Organomegaly within the coelomic cavity will reduce the air sac volume and lead to clinical signs of dyspnoea.

Urinary system

Avian kidneys lie retroperitoneally in the renal fossa of the synsacrum, dorsal to the abdominal air sacs, extending from

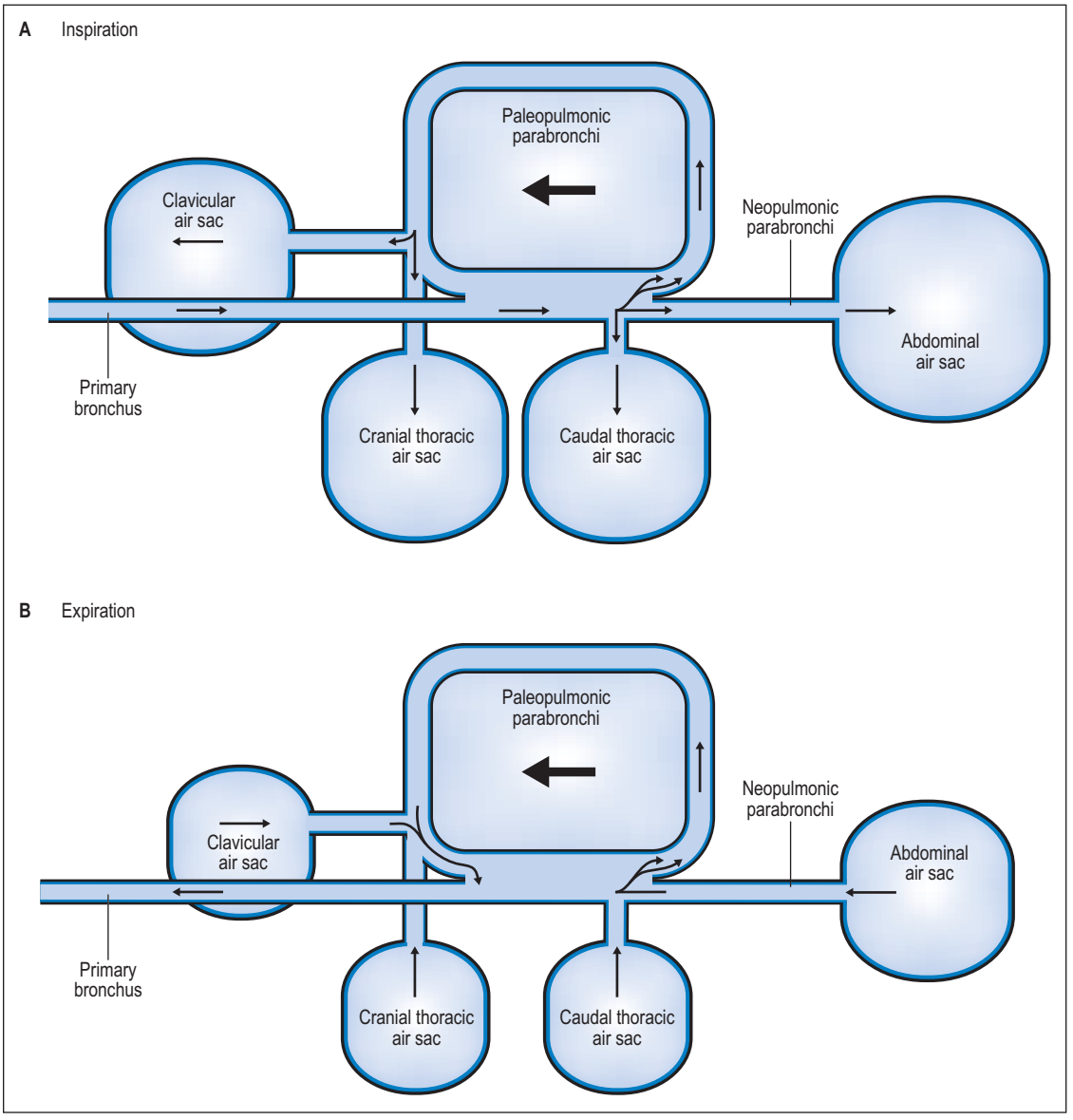


Figure 9.3 • Air flow in the avian respiratory system. (A) Inspiration. (B) Expiration.

the caudal edge of the lungs to the caudal synsacrum (Canny, 1998). Renal disease or enlargement will put pressure on the lumbar and sacral plexus nerves, and clinical signs often include lameness (O'Malley, 2005). Birds have two types of nephron, medullary nephrons (10–30%) that have loops of Henle and are able to concentrate the urine and cortical nephrons (70–90%) with no loop of Henle (Casotti et al., 2000).

There is a dual afferent blood supply to the kidneys (see Fig. 9.2) (Lumeij, 2000). These blood vessels are the cranial, middle and caudal renal arteries, and the caudal renal portal vein (Johnson, 1979; King and McLelland, 1984). The latter renal portal system is important in elimination of urates (Siller, 1971). As mentioned above, a smooth-muscle valve in the common iliac vein controls the amount of venous blood entering the kidneys. When stressed, sympathetic activity stimulates adrenaline (epinephrine) release, opening the valve; this diverts blood from the kidney to the caudal vena cava and thence vital organs such as the heart and brain (Siller, 1971). When the valve is closed, venous flow from the legs enters the kidneys before any other organs (Larochelle et al., 1992). Blood

within the renal portal ring may also bypass the kidneys by shunting to the caudal mesenteric vein or internal vertebral sinus (Echols, 2006).

Arterial blood terminates in the glomerular capillary beds, but renal portal blood is directed to the renal tubules (Wideman, 1988). It is, therefore, possible for blood from the legs to enter the renal parenchyma directly, which may increase the possibility of nephrotoxicity with some drugs administered into the leg (Echols, 2006). Drugs that are eliminated by tubular secretion may also enter this system and be eliminated (Steinhort, 1999). Although the valvular system means this is not always the case, parenteral drugs should not be administered in the caudal half of the body.

Like reptiles, avian kidneys produce both urine and urates; 60% of avian nitrogenous waste is excreted as urates, with urea only formed as a by-product of detoxification in the kidney and in the liver (O'Malley, 2005). Uric acid is synthesised in the liver and excreted mainly by tubular secretion in the kidneys (Goldstein and Skadhauge, 2000; Phalen, 2000). Urate production is independent of urine production and urates will continue to be produced even in dehydrated birds. As with azotaemia in mammals, elevated blood uric acid levels are only seen with severe (70%) renal damage. High serum levels of uric acid or renal damage will lead to gout, with precipitation of uric acid crystal in joints or viscera. Swellings at joints may be visible or palpable (Lawrie, 2005).

Unlike reptiles, birds can concentrate urine, though not as well as mammalian species. It is normal for hens to become polyuric before egg-laying, as diuresis occurs as they excrete excess phosphate released from bone during egg production (O'Malley, 2005).

Birds do not have urinary bladders, and the ureters pass directly from the kidneys to the urodeum of the cloaca. Urine and urates mix with faeces in the rectum and colon, where further reabsorption of water may also occur (Sykes, 1971).

Osmoregulation mechanisms vary between avian species. Most obtain water from food or by drinking (O'Malley, 2005). Desert species have a lower water intake than tropical species, for example a cockatoo may drink less than 50 ml/kg/day while a lovebird can drink 180 ml/kg/day (Wolf and Kamphues, 1992). Nasal or salt glands above the avian eyes are involved in water homeostasis, particularly in birds from arid climates (Evans, 1996; Maina, 1996). Desert species also produce more concentrated excrement than those with more water access and cool warm air in the nares to conserve water (Orosz et al., 1997).

Water homeostasis is performed by renal regulation of electrolyte balance (Wideman, 1988). The regulation of fluid volume results in blood pressure regulation (Echols, 2006). Although avian species can autoregulate the glomerular filtration rate across a wide range of arterial blood pressures, severe hypotension will result in cessation of urine flow (Forman and Wideman, 1999). Birds appear to be able to maintain blood pressure and thereby renal blood flow, in quite severe hypovolaemia (Bottje et al., 1989).

Renal disease is common in avian species and is often due to malnutrition. Excessive dietary protein may lead to accumulation of waste products, uric acidaemia and uraemia; high-cholesterol diets may induce renal disease; and excess dietary lipids will affect the progression of chronic renal disease (Harrison and McDonald, 2006; Klumpp and Wagner, 1986). Hypercalcinosis or hypervitaminosis D may also cause renal disease (Chandra et al., 1985; Phalen et al., 1990). Hypovitaminosis A may also affect uric acid elimination and result in gout (Koutsos and Klasing, 2002; Siller, 1981). Dehydration or fasting is more likely to produce renal, articular or visceral gout than renal disease *per se*.

Various infectious agents, such as bacteria, parasites (for example, coccidian, *Encephalitozoon* spp.) or fungi may result in renal disease (Echols, 2006). Chronic inflammatory disease may result in amyloidosis, which may involve the kidneys (Rigdon, 1974). Toxins such as ochratoxin A, from *Aspergillus* and *Penicillium*, or lead may cause renal damage (Manning and Wyatt, 1984; Mateo et al., 1997). As with other species, renal neoplasia has been reported (Freeman et al., 1999; Latimer et al., 1996). Urolithiasis or ureteral obstruction may occur (Siller, 1981).

Digestive system

The choana is a slit-like opening in the roof of the oropharynx, linking it with the nasal cavity. The anatomy of the avian tongue varies between species (King and McLelland, 1984). The oesophagus lies to the right of the neck (O'Malley, 2005), and most species have a crop, which is a dilation in the oesophagus at the base of the neck.

The digestive system is short and low-volume; hence avian patients feed frequently to supply the energy requirements necessary for their high metabolic rate. Fasting times before anaesthesia should, therefore, be minimised and feeding resumed as soon as possible on recovery. Small birds, such as budgerigars (*Melopsittacus undulates*), should not be fasted. Larger birds are normally fasted long enough for the crop to empty, reducing the risk of regurgitation and aspiration during anaesthesia.

The gastrointestinal system terminates at the cloaca. This is the joint ending from the proctodeum of the urogenital (urodeum) and digestive (coprodeum) tracts (King and McLelland, 1984). The cloaca exits the body at the vent.

Dietary requirements vary between species (Table 9.3). Most psittacines eat a mix of fruit, vegetables and pulses. Species such as lorikeets are nectarivorous, feeding on pollens, plants, insects and their exudates. Raptors are carnivorous, feeding on whole prey. Several species consume insects as part of their diet; in captivity this may include mealworms, earthworms and crickets (McDonald, 2006).

Improper diets fed to captive birds may predispose many systemic abnormalities. All-seed diets are usually deficient in vitamin A. Hypovitaminosis A is commonly seen in passerines and psittacines, and may result in depression of the immune system (Koutsos et al., 2003). Severe hypovitaminosis A may result in gout (Austic and Cole, 1972). An inappropriate high-fat diet often predisposes to obesity, especially in sedentary captive birds. This may lead to hepatic lipidosis and congestive heart

Table 9.3: Dietary requirements in birds

ORDER	PREDOMINANT DIET
Columbiformes	Vegetable matter (fresh green leaves, fruit or seeds); young fed on crop milk
Falconiformes	Carnivorous Variety of whole carcases in captivity, e.g. day-old chick (yolk often removed), quail, rabbit, rat
Passeriformes	Fruit, arthropods, seeds; species variations (some will eat nectar or small vertebrates) Extruded diets are available for pets
Psittaciformes	Vegetable matter (fruit, seeds, buds, nectar, pollen), occasionally insects Extruded diets are available for pets
Strigiformes	Carnivorous (as for Falconiformes)

failure. Epithelial tissues may be affected, increasing susceptibility to respiratory and renal disease. Diabetes mellitus may ensue or be exacerbated by renal dysfunction (McDonald, 2006).

Birds on all-seed diets are often immuno-incompetent. Assume that such birds will have systemic abnormalities and are likely to have infections.

Gastrointestinal transit times may be affected by disease, for example motility disorders or impactions. There are many aetiologies for gastrointestinal motility problems in birds, including bacterial enteritis, proventricular dilatation disease in psittacines and lead toxicity. These may delay crop emptying, which will increase the risk of regurgitation during anaesthesia. Birds with diarrhoea will rapidly become dehydrated, particularly if they are also inappetent (Coles, 1997).

Clinical signs associated with gastrointestinal disease will vary depending on the cause and site within the tract. Signs may include regurgitation, vomiting, diarrhoea or non-specific signs of illness, such as lethargy and reduced appetite. These patients will be malnourished and likely dehydrated; if possible, supportive care should be given to stabilise the bird before anaesthesia is induced. Some conditions will require immediate anaesthesia to allow treatment, for example cloacal prolapse. In these individuals, the risks associated with anaesthesia are increased; fluids and supplemental heating should be provided during anaesthesia and the recovery period.

Although malnutrition is the most common aetiology for *hepatic* disease, infectious or toxic agents may also affect liver function. Clinical signs of hepatic disease are often vague – lethargy, inappetence, polydipsia, polyuria and diarrhoea may be seen. Hepatic dysfunction may be more specifically evidenced by the presence of loose green faeces and green-stained urates (due to the bile pigment biliverdin), ascites, melaena, abnormal beak and nails, or malcoloured feathers. If hepatic fibrosis occurs, it will increase resistance to blood flow (Hochleithner et al., 2006). The caudal mesenteric vein drains the lower gastrointestinal system and may transport disease, such as infection, to the kidneys (Echols, 2006).

Clinical problems in hepatic disorders include coagulopathy and hypoalbuminaemia (Harrison et al., 2006; Hochleithner et al., 2006). Serum bile acids may be elevated (O'Malley, 2005). Aspartate amino transaminase (AST) elevation without creatinine phosphokinase (CPK) increase is suggestive of hepatic damage. Radiography may demonstrate hepatomegaly or microhepatica.

The liver is the site for metabolism of many drugs, including some anaesthetic agents. Hepatic dysfunction will, therefore, adversely affect the patient during anaesthesia and recovery. If hepatic disease is suspected, the patient should be stabilised before anaesthesia; fluids, nutritional support and metabolic aids such as s-adenosylmethionine (SAMe) are advised. If anaesthesia cannot be postponed, an anaesthetic which undergoes minimal hepatic metabolism, such as isoflurane, should be used.

Integumentary system

The skin is very thin in birds and susceptible to injury (Spearman, 1971). Thin membranous regions of skin called patagia are found at various locations in birds, for example the propatagium between the shoulder and carpus, and also where the neck and legs join the body (O'Malley, 2005). These regions are useful sites for subcutaneous injections (Fig. 9.4), including fluid administration.

Feathers assist thermoregulation, by providing insulation for birds. Moulting is triggered by several factors that will be species-dependent. At this time, the bird has increased demands for protein, calcium and iron to form new feathers. As most species moult and replace their feathers only once a year (twice in some species), it is important to avoid damaging feathers during hospitalisation that may affect the bird's performance or flight (see Fig. 11.2) (Bauck et al., 1997).

Reproductive system

As the gonads are located near the kidneys, gonadal enlargements (for example, seasonal reproductive activity or reproductive disease) may affect the lumbar and sacral plexus nerves similarly to renomegaly. Egg production may also

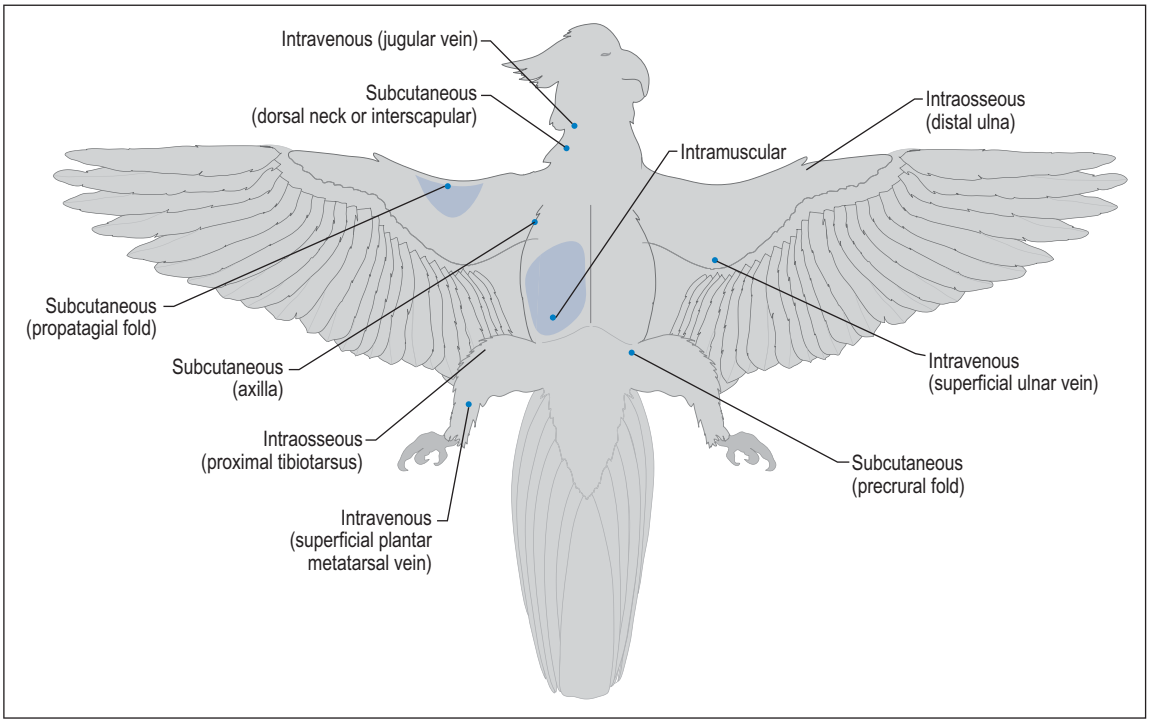


Figure 9.4 • Injection sites in birds.

cause signs of dyspnoea as the abdominal air sacs are compressed by the egg.

Birds with egg binding presented to the veterinary surgeon may require anaesthesia for resolution of the problem. Malnutrition is often a factor in the aetiology of this problem, or it may result from infection, environmental stressors or dystocia. These patients, therefore, often present severely debilitated and benefit from supplemental fluids, nutrition and heat to stabilise their condition before anaesthesia is induced. Analgesia may be necessary.

Cloacal prolapses commonly occur secondary to straining due to egg laying, masturbation or space-occupying intra-abdominal lesions (Bowles, 2006). These prolapses may contain sections of reproductive, digestive or urinary tracts. Anaesthesia is usually required to treat prolapses. As the underlying aetiology of the prolapse is likely to be due to or have caused some degree of systemic compromise, supportive care is paramount for the successful anaesthesia of these patients.

Endocrine system

Corticosterone has both glucocorticoid and mineralocorticoid activity in birds and is, therefore, important in electrolyte balance (O'Malley, 2005). In birds, glucagon from the pancreas is more important in carbohydrate metabolism than insulin. Diabetes may occur in some birds, for example in cockatiels.

Nervous system

Central nervous system disease in birds may be due to several aetiologies. These include infection, toxins, trauma, neoplasia and dietary deficiencies (vitamins B or E, calcium) (Coles, 1997). After initial assessment and stabilisation, patients that have suffered head trauma should be placed in a quiet, darkened enclosure with minimal stimulation (Harrison et al., 2006).

Special senses

Vision and hearing are the most important senses in birds and avian species should be hospitalised in a quiet darkened area of the veterinary practice to reduce stress.

Systemic disease

Commonly, birds presented to the veterinary surgeon and requiring anaesthesia will be debilitated due to poor diet and/or environment. Immune depression may result in generalised infection in the avian patient.

PRE-ANAESTHETIC ASSESSMENT/STABILISATION

History and clinical examination

Triage should be performed during the initial assessment of the patient. At any point during this phase the veterinary clinician should be prepared to pause and postpone investigative techniques, in order to perform immediate treatment to critical patients or euthanasia as deemed necessary. As most pet birds are presented with some degree of illness, there is always the possibility that the patient may die when stressed during investigation or treatment. The bird's owner should be kept informed at all times, particularly on the likely outcome of each procedure. Initial assessment of the patient should identify signs of illness and experience will allow the veterinary surgeon to make a clinical judgement as to the appropriate action.

As with other exotic species, a full *history* should be taken before examining the bird. This will include previous and current husbandry conditions, including diet, contact with other birds or toxin exposure, and any prior or current medical problems. This part of the investigation is extremely important to identify any risk factors for disease; birds will mask signs of illness until disease is severe and clinical signs are usually subtle, often making the clinical examination inconclusive. Owners that have strongly bonded birds are more likely to notice signs early on in disease.

Observation of the bird from a distance after it has settled in the consulting room will allow the clinician to assess for clinical signs of illness, including assessment of demeanour, paresis or neurological abnormalities, respiratory function and identification of dyspnoea if present. A bird showing clinical signs is likely to be severely ill. The bird may be able to hide signs for a short period when initially brought into the surgery, and it is beneficial to postpone direct assessment of the bird by discussing husbandry and history details with the owner first. If the veterinary surgeon suspects the presence of disease based on the history, or if signs of illness are observed, the owner should be advised of the risks associated with restraint and clinical examination of their bird.

A full *clinical examination* is possible with most birds, although should be brief in those with a history of respiratory problems or exercise intolerance. Most species are restrained using a towel, controlling the wings and legs to prevent injury, but allowing keel movements during respiration. This may lead to hyperthermia if restraint is prolonged, particularly in obese birds, and the patient should be closely monitored. For birds of normal body weight panting will commence after wrapping in a towel for 5 min, but the time will be less for overweight individuals and the examination should be briefer for these cases. Birds that are not used to handling may be fatally stressed by restraint; the owner should be forewarned of this possibility.

The pre-anaesthetic assessment should include examination of the nares, oral cavity, choanal slit and glottis, along with gentle palpation of the coelomic cavity. Cardio-respiratory function can be further assessed by auscultation of the heart, lungs and air sacs. Any injuries present should be related to avian anatomy, for example bone fractures in the cervical or humeral region may affect the respiratory system by puncturing the cervical or clavicular air sac. Subcutaneous emphysema may also be seen if pneumatised bones are fractured. External abnormalities often relate to internal disease that may increase the risks of anaesthesia, for example malnutrition or hepatic disease may lead to poor feather quality.

Palpation of the pectoral muscles and the keel prominence will allow the clinician to assess the bird's condition. Excess fat may be laid down subcutaneously over the pectoral muscles and can be visualised if the feathers are dampened with an alcohol-soaked swab. An accurate body weight should also be obtained, to help assess body condition and to permit accurate dosing of drugs. The patient may be weighed in a plastic box on digital scales (see Fig. 1.9) or in a cloth bag using a spring balance. Some trained birds will sit on a perch placed on top of scales. It is not advisable to guess the weight of a bird prior to drug administration, as a small volume increase may be a fatal overdose. Scales also enable the clinician to assess a patient's progress, with daily weighing part of the clinical examination of hospitalised animals and intermittent weight assessment important for bird check-ups.

The cardiovascular system can be assessed during the pre-anaesthetic clinical examination as well as monitored during anaesthesia. Capillary refill can be assessed on non-pigmented skin over the feet and should be less than 1 s. Refill time after digital pressure on the basilic or ulnar veins at the medial elbow should also be less than 1 s if perfusion is normal. Cardiac auscultation is performed over the pectoral muscles. Heart rates are extremely variable between species, usually between 110 and 300 beats per minute. Smaller birds generally have a higher rate, although stress and excitement will increase heart rate significantly in all birds. Difficulty in hearing heart sounds may occur if the patient is stressed, or if pericardial fluid or soft tissues surround and insulate the heart. Murmurs and arrhythmias are more easily appreciated in quiet individuals or under general anaesthesia. Sinus arrhythmia is normal in avian species (Raftery, 2005).

Although auscultation of the respiratory system is not always productive, any wheezes, crackles, clicks or squeaks heard are likely to indicate disease. An infant or paediatric stethoscope should be used to listen bilaterally both dorsally and laterally over the rib cage, and ventrally over the coelomic cavity. The stress of the physical examination may cause the bird to show more clinical signs of disease than when relaxed and compensating. This is particularly the case with cardio-respiratory disease. Birds with disease often hide signs of illness, so the owner may not have noticed any clinical signs, but the stress of handling may destabilise the patient and produce clinical signs, such as open-mouth breathing. The time taken to return to normal can be a useful indicator of the severity of disease. If concerned about a dyspnoeic patient, the patient should be returned to its cage or into a chamber where oxygen can be supplemented. If more than 3 min is required for

the bird's respiratory rate to normalise, respiratory capacity is reduced (Raftery, 2005). Dyspnoeic birds should be placed in an oxygen chamber for stabilisation before a complete physical examination can be performed. However, if an upper airway obstruction is suspected, anaesthesia and air sac cannulation are indicated.

Renal function and disease can be assessed using blood and urine analysis, and imaging techniques. However, renal biopsy is required to diagnose renal disease definitively. The initial history and investigation may alert the clinician to a possible renal problem, but anaesthesia will be required for a biopsy to be performed. The choice of anaesthetic protocol should consider problems with renal dysfunction, such as a reduced glomerular filtration rate, likely uricaemia, and possible systemic disease. Fluids should be administered before, during and after anaesthesia to correct and maintain hydration, and to maintain systemic blood pressure and renal blood flow. Drugs that may be metabolised or excreted by the kidneys should be avoided in such patients.

It can be difficult to diagnose the exact cause of an avian patient's illness, but triage should help decide whether it is necessary to perform further investigation. In some cases it may be more appropriate to postpone these and initiate supportive care to improve the bird's condition before it is stressed any more. If the patient is very thin, it should not be stressed by excess restraint; pre-existing hypoglycaemia in emaciated patients may result in sudden death when the metabolic demand is increased. If hypoglycaemia is suspected, gavage-feeding or intravenous administration of dextrose solutions or glucose should be performed.

Examination of fresh droppings may allude to specific gastrointestinal problems or more generalised disease. For example, the presence of green biliverdin-stained urates suggests hepatic dysfunction, which may increase the risk associated with anaesthesia. It is important to have knowledge of the normal appearance of droppings for various species, as different dietary constituents will affect the droppings. A faecal Gram stain assessing microflora may demonstrate gastrointestinal abnormalities.

Brief ultrasonography may be possible consciously in some birds. Further *investigations* in patients often require sedation or general anaesthesia, as restraint for prolonged procedures is stressful and potentially fatal, particularly in smaller birds. The owner should be advised of the risk associated with general anaesthesia in avian species, along with any specific risks for the pet.

Phlebotomy may be performed in conscious birds, but restraint is usually stressful during the procedure, as well as after to apply haemostatic pressure. A small blood sample will allow the clinician to perform several cage-side tests. A microhaematocrit capillary tube will allow assessment of the haematocrit (packed cell volume [PCV]), which is normally between 35% and 55% in most avian species, and an estimate of total solids using a refractometer (Coles, 1997). A blood smear can be used to estimate total white cell count and white cell differential, as well as show an anaemia or thrombocytopenia. Glucometers can be used to check for hypoglycaemia or, more rarely, hyperglycaemia. These parameters can be an invaluable part of the patient's assessment and are especially useful when the clinician must wait for an outside laboratory to return results on other blood analyses. An elevated PCV (>55%) and hyperproteinaemia may suggest dehydration and fluid therapy should be initiated. If the bird is anaemic, with a PCV <20%, a blood transfusion should be administered. If the bird is obviously unwell and anaesthesia can be postponed pending results, a larger blood sample should be analysed for other haematology and clinical chemistries. If this delay is necessary, it allows the clinician to stabilise the patient to improve the bird's condition before anaesthesia.

Hospitalisation facilities

Birds are susceptible to *stress* and care should be taken during hospitalisation to reduce any stressors on the patient. This will include attention to the patient's environment, including positioning the bird's cage in a quiet area of the veterinary practice, out of sight of larger predator species, such as dogs and cats. Similarly, predator bird species should not be within sight of other birds; hence passerines and psittacines should be kept separate from birds of prey. Dimming the lights or partially covering the cage to reduce the light intensity will also be less stressful than the glare of artificial lights in veterinary practices. If possible, the bird should be allowed a period of time to acclimatise to the hospital environment before anaesthesia is induced. Certain birds will become very stressed in a new environment and in some cases may benefit from a minimum period in the veterinary practice before anaesthesia is induced.

If a contagious disease is suspected the bird should be maintained in a separate airspace or, ideally, have a separate air *ventilation* system. Handling should be minimised, particularly if the individual is not habituated. Species and individual differences will affect how easily the bird adapts to hospitalisation. Companion birds often benefit from human activity and voices in the ward or a radio may be left on to mimic normal home noise. Most birds are more stressed if kept at a low height and so should not be kept at floor level.

Most birds will not accept new *diets* readily. It is, therefore, important either to stock a variety of foods or to request that the owner provides some of the patient's usual diet. If the practice does not stock or is unable to purchase the bird's usual food, most owners will be happy to bring some in for the patient's hospital stay. Birds have high metabolic requirements, particularly passerines (Lasiewski and Dawson, 1967). It may be easy to see if a Harris' hawk (*Parabuteo unicinctus*) has eaten a chick, but less easy to assess whether a budgerigar (*Melopsittacus undulates*) has eaten or merely de-husked seeds. Other birds may refuse to eat even their normal diet within the new and stressful environment of a veterinary practice. In order to monitor food consumption and assess weight gain or loss, hospitalised birds should be weighed at the same time once daily. Keel prominence should also be checked at this time, as pectoral muscle mass is a useful indicator of body condition. If poor condition or weight loss is noted, supplemental nutrition should be administered, usually in the form of gavage feeding (Table 9.4).

Table 9.4: Fluid and nutritional support for birds

FLUID	ROUTE	DOSE	FREQUENCY	COMMENT
Blood[2]	IV	10–20% of bird's total blood volume	Dependent on severity of anaemia, aetiology of disease, type of transfusion	Homologous donor preferred; heterogeneous species donor acceptable for single transfusion
Colloids, e.g. gelatin artificial colloidal substitudes with electrolytes, hetastarch[4]	IV	10–15 ml/kg[5,7] 5 ml/kg + 15 ml/kg lactated Ringer's[4]	q8h, up to 4 treatments	Volume expansion Treatment of hypovolaemia, hypoproteinaemia (total solids <20 g/l) Can be used in anaemic patients Caution in patients with congestive heart failure or renal failure
Haemoglobin glutamer-200 (bovine) (Oxyglobin®, Biopure Netherlands BU)[4]	IV	30	Once	Divided doses, over 24 h
Crystalloids:		<10 ml/kg/h	CRI	Routine administration during anaesthesia
		50 ml/kg/day[6]	Bolus	Maintenance fluids
Hartmann's solution 5% Glucose + 0.9% saline	IV, SC, IO IV, IO			
25% dextrose	IV	1–2ml/kg[3]	Slowly, to effect	Treatment of hypoglycaemia
Electrolyte solutions, e.g. Lectade® (Pfizer Ltd, Kent, UK)	PO	50 ml/kg/day[6]	Divide into 2–4 doses	Oral rehydration and electrolyte replacement
Nutritional support with amino-acids and essential vitamins: Duphalyte® (Fort Dodge Animal Health, Southampton, UK) Critical Care Formula® (Vetark, Winchester, UK)	SC PO	10 ml/kg[1] 5 ml scoop with 10 ml water/100 g/day	q6–8 h Divide into 2–3 doses daily	
10% Glucose or glucose saline[1]	PO	10	q6h	Will provide supplemental nutrition, but not cover maintenance requirements
Harrison's® Recovery Formula (Harrison's Bird Foods, http://www.harrisonsbirdfoods.com/)	PO	5 ml/100 g bird	Repeat 1–4 times daily.	Nutritional or support formula for sick or injured birds
Canine/feline a/d® (Hill's, Herts., UK)	PO	<5 ml/100 g		Can dilute with water Carnivore convalescent diet

Key: CRI, continuous rate infusion, IO = intraosseous, IV = intravenous, PO = oral, SC = subcutaneous, q6h = every 6 hours

[1] (Coles, 1997); [2] (Degernes et al., 1999a; Degernes et al., 1999b); [3] (Harrison et al., 2006); [4] (Lichtenberger, 2004; Orcutt, 2000); [5] (Morrisey, 1997); [6] (Raftery, 2005); [7] (Stone, 1994)

The basal metabolic rate will also increase during disease, as the inflammatory response increases energy demands (Coles, 1997). Energy demands will be reduced if the environmental *temperature* is raised to 26–29°C. This can be done most simply by keeping the ill bird in an incubator (Fig. 9.5) or brooder (Fig. 9.6), which will have a thermostatically controlled temperature. Most incubators also allow oxygen supplementation, which may be useful in dyspnoeic patients or during recovery from anaesthesia. It is also useful to provide humidity of around 70%, either with a humidity source in the incubator or by placing a small dish of water or a damp towel in the enclosure (Harrison et al., 2006). With larger birds, infrared heat lamps or hot water bottles wrapped in towels can be used to increase the patient's local temperature. The cage should be protected from draughts. A digital thermometer should be used to monitor the temperature and ensure overheating does not occur.

All birds appreciate somewhere to *perch*; this also protects their tail feathers by keeping them above the substrate. The type of perch required will depend on the species and varies from wooden dowelling to more solid block perches. Whichever type of perch is used in the veterinary practice, it should be easy to disinfect between patients. Temporary perches can be made from a variety of materials, such as upturned plant pots or non-toxic branches. It may be helpful to provide a couple of different types of perch, so the patient can select one or move around. The perch should be placed low in the cage for weak or uncoordinated birds, or those recovering from anaesthesia.



Figure 9.5 • Incubator for use with small exotic pet patients (AICU® [Animal Intensive Care Unit], Animal Care Products, various suppliers).



Figure 9.6 • Brooders are useful during recovery from anaesthesia or for hospitalisation of critical patients (Brinsea TLC-4M Intensive Care Unit®, North Somerset, UK).

BOX 9.1 Basic equipment required to hospitalise birds

- Digital weighing scales
- Quiet environment
- Escape-proof cage
- Perches
- Bowls for food and water
- Gavage feeding tubes
- Diets appropriate for various species, e.g. fresh fruit and vegetables, seed mix, parrot formulated diet and day-old chicks (can be stored frozen)
- Nutritional support diets
- Handling: towels, falconer's glove
- Incubator/brooder: for heat, humidity, and oxygen provision

Stabilisation

Most birds presented at the veterinary practice will be unwell, with fluid and/or nutritional imbalances that may adversely affect the patient during anaesthesia. The exception would be the patient that has undergone acute trauma, but even in these cases pain or blood loss may lead to shock that should be treated before anaesthesia is induced. Most birds, therefore, benefit from a period of stabilisation before

being anaesthetised. This period may vary from 1 h to 2 days, depending on the clinician's initial findings. Each case will be different, but the veterinary surgeon should decide whether to perform a multitude of tests and procedures at one time, reducing the handling frequency, or to limit treatments to one procedure before the bird is returned to a cage to recover from handling before another procedure is performed.

Oxygen supplementation

Patients presenting with signs of dyspnoea or suspected cardiovascular disease benefit from supplemental oxygen; the least stressful method of administration is an oxygen chamber with 40–50% oxygen (Quesenberry and Hillyer, 1999). If a chamber is not available, a clear plastic bag can be used to cover the bird's cage or carrier to contain oxygen supplied from an anaesthetic circuit. The use of a facemask may cause stress as restraint of the bird is usually necessary. Prolonged high concentrations of oxygen should be avoided as they may result in oxygen toxicity (Harrison et al., 2006). Humidifying the oxygen (to at least 60% relative humidity) will also reduce dehydration due to loss from respiratory passages (Pees et al., 2006). If the bird's condition deteriorates or the dyspnoea does not reduce within 30 min, cases with upper airway obstruction should have their air sac cannulated (Raftery, 2005).

Fluid therapy

Most birds obtain water from metabolised food and body fat stores. Therefore, dehydration will occur rapidly when birds are ill and anorexic. Although birds will retain water at these times by excreting less urine, they will benefit from rehydration. All fluids should be warmed to body temperature, approximately 39°C, to reduce the risk of hypothermia following administration (Coles, 1997).

The daily water intake for most birds is 40–60 ml/kg (Curro, 1998; Steinhort, 1999), so maintenance fluids are provided at 50 ml/kg/day. In dehydrated patients, the deficit should be estimated and replaced over a period of approximately 24 h (Raftery, 2005).

> Hydration should be corrected in ill birds before gavage feeding is commenced.

The easiest route for rehydration is administration of fluids orally, usually by crop or gavage feeding. Nutrition can be provided concomitantly when required. This route is ideal for patients with normal gastrointestinal function and consciousness, as the major risk associated is aspiration after regurgitation. Up to 20 ml/kg can be administered orally and is usually repeated every 6–8 h. If required, other routes can be used additionally to provide fluids.

Subcutaneous fluids are relatively easy to administer, but absorption is slow and suitable sites restricted in birds. Severely debilitated or dehydrated patients will not absorb subcutaneous fluids (Harrison et al., 2006). Some texts recommend the addition of hyaluronidase (150 IU/L of fluids) to increase the rate of absorption from subcutaneous fluids (Griffin and Snelling, 1998). Fluids should be warmed before administration to reduce the risk of hypothermia.

For birds with hypovolaemia and others with severe dehydration, intravenous or intraosseous fluids are used to result in rapid rehydration. However, these routes do produce some stress to the patient for their delivery and anaesthesia may be required for placement of catheters. Intraosseous access is ideal for very small or collapsed patients where venous access is not possible. It is more painful to insert a needle intraosseously, but can be lifesaving in shocked patients. The intraosseous route is analogous to intravenous, entering the circulation, and fluids or drugs that may be administered intravenously can also be given intraosseously. Placement of a venous or intraosseous catheter is discussed later (see Techniques section).

Crystalloids are useful first-line fluids to treat dehydration in avian patients. Glucose-saline is useful as the glucose provides a source of carbohydrate and the saline replaces extracellular and intracellular fluid to expand the circulating fluid volume temporarily. Lactated Ringer's solution provides electrolytes and fluid. The lactate undergoes hepatic metabolism to produce bicarbonate that helps correct acidosis (Raftery, 2005).

Colloids may be synthetic or natural and are used to expand plasma volume in hypovolaemic shock. When used in combination with crystalloids, the volume required is reduced by 40–60% (Raftery, 2005). If severe anaemia is present, a colloid with oxygen-carrying ability (for example Oxyglobin®, Biopure Netherlands BU) should be chosen. In other instances, synthetic colloids (for example Haemaccel®, Intervet UK) can be used; these are cheaper and more readily available.

Patients with hypovolaemic *shock* will generally have pale mucous membranes, slow venous refill time, elevated heart rate and falling haematocrit (Lichtenberger, 2004). A slow bolus of lactated Ringer's solution (10 ml/kg) combined with haemoglobin-based oxygen carrier or hetastarch (5 ml/kg) should be administered over 5 min (Raftery, 2005).

In many cases where fluids are required, the bird is bright enough to entangle itself or attack a fluid giving set and it is easier to place a bung in the catheter to administer boluses. In debilitated patients, a continuous rate infusion may be more appropriate. Bolus fluids may result in hypervolaemia and polyuria, while a continuous infusion will allow retention of more fluid. Syringe pumps can be used to administer very small volumes, for example 1 ml over 1 h. Hypoproteinaemia (total solids <20 g/l) can be treated using intravenous administration of hetastarch (Harrison et al., 2006).

The administration of boluses allows the clinician to provide fluids intravenously or intraosseously at periods throughout the day. Maximum recommended volumes of intravenous fluid boluses are 1.0 ml for a budgerigar (*Melopsittacus undulates*), 2.0 ml for a cockatiel (*Nymphicus*

hollandicus), 8.0 ml for an Amazon parrot (*Amazona* sp.) and 12 ml for a large macaw (*Ara* sp.) or large cockatoo (*Cacatua* sp.) (Coles, 1997). As with other medications, the veterinary surgeon must balance the stress caused to the patient by repeated handling and restraint against the benefits gained from administration of fluids.

Fluids may be administered rectally if no other routes are available. Absorption is good via this route, but it is imperative to warm fluids to avoid hypothermia. Care should be taken during administration not to damage the delicate recto-cloacal tissue.

Birds in *renal failure* should be given 10% of their body weight in fluids daily (Lumeij, 2000). Warmed fluids are administered orally, intravenously or intraosseously; 50–100 ml/kg is administered twice daily to diurese the patient (Echols, 2006). Balanced electrolyte solutions are appropriate. Allopurinol may be used to reduce uric acid production, but renal toxicity has been reported in at least one species (Czarnecki et al., 1987; Lee and Fisher, 1972). A balanced diet should be fed to the patient. Although non-steroidal anti-inflammatory drugs (NSAIDs) may be beneficial in reducing inflammation in renal disease, most are eliminated via renal clearance and have been associated with renal lesions in birds (Klein et al., 1994; Radford et al., 1996). Low-dose aspirin has been used in some birds with renal disease (Echols, 2006).

Anaemia from acute blood loss is particularly concerning in a malnourished individual, as coagulopathy may be associated with hepatic dysfunction. Anaemic patients should be stressed as little as possible, as the increased oxygen demand may be too great for their small oxygen reserves. Oxygen supplementation will increase the efficiency of the remaining erythrocytes (Harrison et al., 2006). Severely anaemic birds (PCV <20%) may require a blood transfusion. Birds with chronic anaemia will adapt, but intervention is required when PCV drops below 12% in these cases (Raftery, 2005).

Blood donors are ideally homologous, that is, from the same genus and species (Degernes et al., 1999a; Degernes et al., 1999b). Reports suggest that a single transfusion from a heterogeneous species is acceptable, but erythrocytes will be short-lived (Harrison, 1984). The donor should be examined for signs of illness and the blood examined for haemoparasites. Subsequent heterogeneous transfusions will produce reactions (Coles, 1997). The transfusion should be 10–20% of the bird's blood volume (Harrison et al., 2006), equivalent to approximately 1% of the bird's body weight. Blood is collected into acid citrate dextrose, with 0.15 ml anticoagulant per ml of blood. Agglutination and haemolysis should be assessed before repeat transfusions are administered, by mixing donor and recipient red blood cells and serum on a microscope slide (Raftery, 2005).

If blood is unavailable, an alternative is haemoglobin glutamer-200 (bovine) (Oxyglobin®, Biopure, Cambridge, MA); where available, this is expensive. Gelatin artificial colloidal substitutes with electrolytes can also be used in anaemic patients to expand the blood volume (Lichtenberger et al., 2001). Erythropoietin has been used in avian patients to stimulate red cell production, at 100 IU/kg subcutaneously three times weekly (Degernes, 1995).

Nutritional support

Birds on a marginal nutritional plane, including hunting birds of prey, may require supplemental nutrition in the peri-anaesthetic period. Dehydration may affect nutrient absorption and rehydration therapy is usually administered initially before commencing nutritional support. If a bird is obviously thin, the addition of glucose to the fluids or the use of a diluted nutritional support formula may be advisable. Proprietary brands of support formula exist, although human baby foods may be appropriate in an emergency – attempting to provide an equivalent to the usual food, for example fruit-based for an Amazon parrot (*Amazona* sp.). The food administered should also be easily absorbed with minimal digestion, for example a carnivore convalescent diet can be fed to birds of prey.

Hand-rearing formulas can be used to treat debilitated adult birds. If they are strong enough and willing to take the food, sick birds can be hand-fed the formula from a spoon. They will require 30–60 ml/kg per day, divided into three or four doses through the day. Convalescent diets have been formulated to provide easily digestible sources of nutrition. The volume that can be gavage-fed at one time will depend on the size of the bird, with adult bird crop capacity approximately 3% of their body weight, for example 3 ml in a 100 g bird. Sick birds may be prone to regurgitation and aspiration, and smaller volumes are usually administered. At this point in time, total parenteral nutrition is not a feasible option for avian species, due to technical and financial constraints (Harrison et al., 2006). The reader is referred to other texts for further information on different nutritional supplements.

Supplemental heating

Anorexic birds are extremely susceptible to hypothermia. Incubation of the patient in an environment warmed to 25–30°C will reduce metabolic demands for thermoregulation. The bird will then be better able to utilise nutrition for other metabolic processes.

Preparation for anaesthesia

Once the patient has been stabilised, preparations can begin for anaesthesia. All equipment for the procedures to be performed should be prepared, to limit the anaesthetic period. Emergency equipment and drugs should also be readied.

EQUIPMENT REQUIRED

Anaesthetic equipment

Anaesthetic *circuits* used will vary depending on the size of the bird. Systems should ideally have a low dead space,

low resistance and be non-rebreathing. For most avian species seen in veterinary practice, a circuit such as a modified Rees, Ayre's T-piece, or Bain may be used. Open or semi-open circuits are necessary, as the tidal volume of birds is insufficient to move gases through closed systems. Relatively high gas flow rates are required to expel carbon dioxide and ensure rebreathing does not occur (Coles, 1997). Oxygen flow rates should be two to three times the minute ventilation with non-rebreathing circuits or 150–200 ml/kg/minute (Edling, 2005).

Non-rebreathing circuits have the advantage of providing almost instantaneous changes in gas provided to the patient after vaporiser adjustment, which is extremely useful in avian patients that have a rapid physiological response to anaesthetic gases. The addition of a reservoir bag in the Rees' modification of the Ayre's T-piece (see Fig. 1.1) permits intermittent positive pressure ventilation (IPPV) to be performed in intubated patients. A bag can also be attached to the Bain. The coaxial mini-Bain (see Fig. 1.2) has the added benefit of warming inhaled gases. It is advisable to humidify inspired gases if possible to reduce water loss from the avian air sacs (Coles, 1997).

As discussed in the 'Techniques' section, uncuffed *endotracheal tubes* (see Fig. 1.6) are required for avian species. Various sizes should be purchased for use in birds, with small sizes available (for example, from 1.5 mm diameter). Shouldered tubes are useful to close the glottis, reducing waste gas contamination of the environment and enabling mechanical ventilation to be performed. For very small birds, intravenous catheters (with the stylet removed) can be attached to a small connector. Rubber feeding tubes or urinary catheters can also be shortened and used.

Most new endotracheal tubes require shortening before use in small avian species. Ideally the tube should be positioned within the trachea with the connector at the beak. As birds already have a large dead space volume within their trachea, it is important to reduce dead space within the anaesthetic circuit. If endotracheal tubes are overly long, they should be shortened proximally, retaining the rounded bevel tip. Similarly, if other tubing is shortened, the end should be cut obliquely and smoothed in a flame to reduce tracheal damage during insertion.

Facemasks are used for anaesthetic induction of many species and also for maintenance of anaesthesia in species that cannot be intubated. The mask should ideally be comfortable for the patient, allow visualisation of the head to assess anaesthetic depth, contain minimal dead space around the patient's head that may increase rebreathing of waste gases and create a seal to prevent contamination of the environment with anaesthetic gases. A variety of sizes are required for various species (see Fig. 1.7). Masks with clear plastic are ideal, as they allow some assessment of the bird, such as eye movement and position.

Many manufactured masks have a rubber diaphragm that fits around the patient's neck, but this may not create a good seal for small birds. For these patients, a larger mask can be adapted to reduce escape of waste gases into the environment by tying an examination glove over the end and piercing it to create a small hole for the patient's head (see Fig. 1.7B). However, if this creates a mask with a large dead space it should not be used for prolonged procedures due to the risk of hypercapnia. The seal created around the bird's neck should be tight enough to permit PPV, but not occlude superficial blood vessels (Edling, 2005). Syringe-cases can be adapted to attach to connectors and used as masks for small birds.

Passive scavenge of waste gases may allow a substantial portion of escape into the environment if the facemask is loosely fitted. It is preferable to use active scavenge if a facemask is used throughout anaesthesia.

Air sac cannulae can be purchased or adapted from existing endotracheal tubes (Fig. 9.7). The basic design is a short tube with an adapter for connection to an anaesthetic circuit, with side-openings in the tube to reduce the risk of blockage from intracoelomic material or viscera. It is helpful to have a rubber disc or other attachment near the connector for suturing the tube to the patient. In an emergency, placement of any piece of tubing would allow access for room air ventilation.

Suction may be required to remove regurgitated fluids or excess secretions that may block the airways during anaesthesia or recovery. Syringes or other suction device should be available.

Anaesthetic monitoring equipment

Equipment is similar to other species, with a competent and attentive anaesthetist essential to monitor the patient's status throughout the procedure. Bell or, in larger patients, oesophageal stethoscopes can be used to monitor heart rate. ECGs can be attached to birds using adhesive pads (Fig. 9.8). Indirect blood pressure monitors can be used. Capnographs are useful in avian species, pulse oximeters less so.

Hospitalisation requirements

Digital *scales* or a spring balance are vital for avian medicine, to obtain accurate body weights for patients.

RELEVANT TECHNIQUES

Routes of administration

This may pose some problems in small birds, as calculated doses may be very small volumes. For some medications, it may be necessary to dilute the agent, for example with sterile water for injection, before administration to avoid overdosage. The use of insulin syringes also improves accuracy.

Oral

The oral route is the easiest method of administering fluid or medications to avian patients and is often a procedure that clients may perform comfortably at home. Medication administered via food or drinking water is variably accepted, particularly in ill patients, and is not advisable if

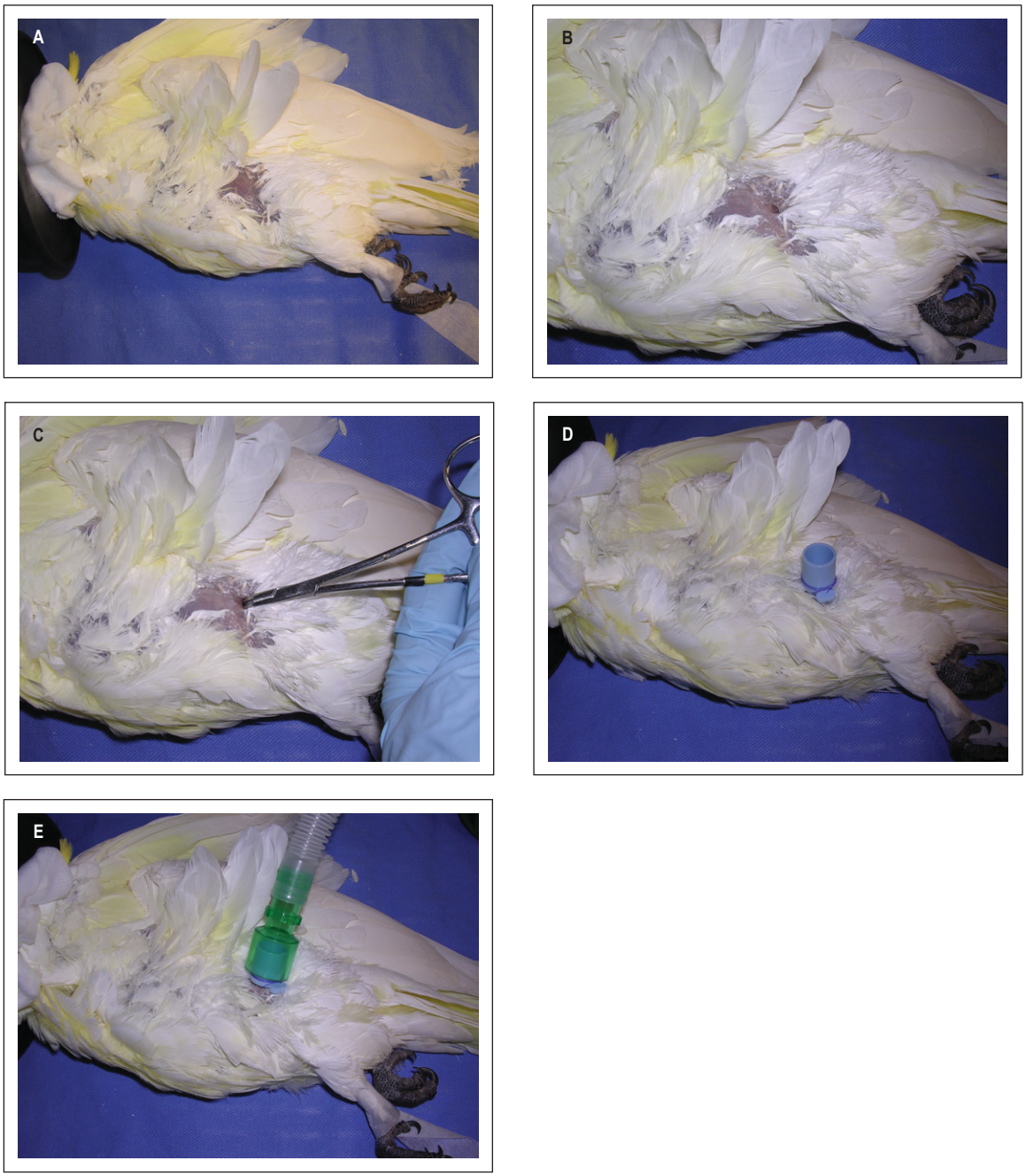


Figure 9.7 • Air sac cannulation. (A) Patient in lateral recumbency, legs held caudally and wings dorsally; skin surgically prepared. (B) Small skin incision caudal to last rib. (C) Forceps in incision to puncture musculature. (D) Tube placed into the caudal thoracic air sac. (E) Anaesthetic circuit can be attached to the cannula.

an exact dose is required. Small volumes can be syringed directly into the oral cavity, but gavage feeding may be required for larger volumes.

Gavage feeding is an easy procedure in most birds. Proprietary crop tubes are available (Fig. 9.9), including metal or plastic (Dosing catheter®, Harvard Apparatus Ltd, IMS, Kent, UK) tubes. Red rubber tubing or lengths of fluid giving sets with smoothed ends can also be used. Care should be taken with the softer tubes not to allow the bird to bite it, as a broken piece of tubing is easily



Figure 9.8 • Electrocardiogram pads can be attached to the skin of birds in similar positions to other species (for example, the right and left wing on the propatagium and left pre-crural fold as seen in this Socorro dove [*Zenaida graysoni*]).

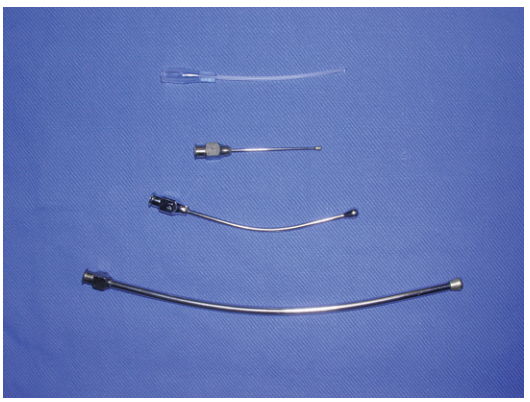


Figure 9.9 • Avian crop tubes may be metal or plastic, and come in a range of sizes.

swallowed by the patient and may require anaesthesia for retrieval. An assistant should hold the beak open using the fingers or bandage material in small patients, or a metal speculum in larger birds, to protect soft tubes. A relatively large tube is selected, to reduce the risk of glottal entry and accidental airway dosing. Most proprietary tubes have a dilated or 'ball' tip to reduce the risk of glottal insertion or oesophageal trauma.

The conscious bird is restrained using a towel, with the head controlled. The head and neck should be extended to minimise the risk of trauma during tube insertion. The lubricated tube is passed lateral and dorsal to the glottis as far as the base of the neck (into the crop in most species or mid-oesophagus in those birds that do not have a crop) (Fig. 9.10). If resistance is felt, the tube should be withdrawn and the procedure re-started. The tip of the tube

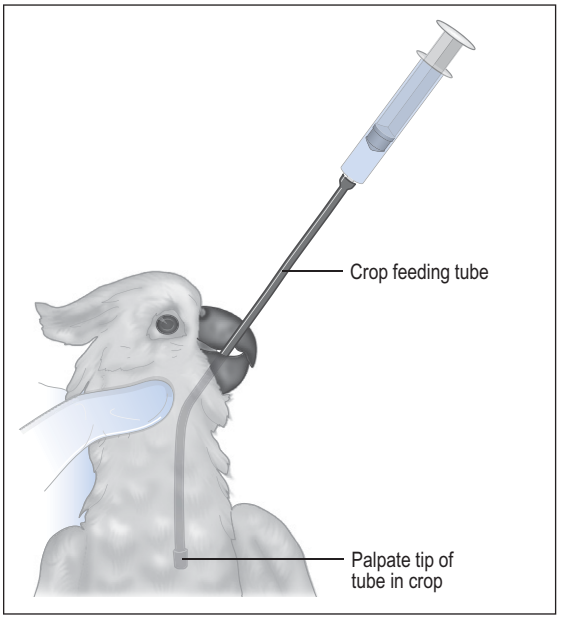


Figure 9.10 • Crop tubing the avian patient. The head and body are restrained while the crop tube is inserted into the mouth. A gag may be useful in some birds. The tube passes dorsal to the tongue, lateral to the glottis.

should be palpated in the crop or oesophagus at the base of the neck before administration of medication.

Fluids and food should be warmed before administration to reduce the risk of hypothermia. Care should be taken not to overheat fluids as internal burns may occur. Up to 20 ml of fluids per kg body weight can be administered with one dose, but less is given to debilitated patients that are more likely to regurgitate; this bolus can be repeated once the crop empties (Chitty, 2005). The bolus should be administered slowly to reduce the risk of regurgitation and the oral cavity observed for this during the procedure. If regurgitation is noted, the tube should be removed to allow the bird either to swallow the medication or release it from its mouth. If the patient is quite debilitated it is often helpful to hold the bird in a head-down position to allow excess fluid to drain and reduce the risk of aspiration. If in doubt, use suction and/or cotton buds to remove fluid from the oral cavity to protect the airway.

Injections

Small-gauge needles are used for injections or catheterisation in birds, as there is a high risk of leakage from blood vessels or skin if larger needles are employed. In most cases 25-gauge needles are sufficient, but 27-gauge may be more appropriate for very small and 23-gauge for larger patients.

Intramuscular injections are usually administered in the pectoral muscles (see Fig. 9.4). The injection should be into the mid to caudal portion of the muscles, as the

veins within the muscles are larger cranially (Coles, 1997). A needle of 23-gauge or smaller is usually used, depending on the viscosity of the medication and the size of patient (Chitty, 2005). The syringe plunger should be withdrawn as with other injections to ensure a blood vessel has not been inadvertently entered. The iliotibialis lateralis or biceps femoris muscles may be used in larger birds, avoiding the sciatic nerve posteriorly; the renal portal system may drain the drug directly to the kidneys from this site. If large volumes (greater than 1 ml per 500 g body weight) are to be administered, the injection should be divided between two or more sites to reduce the risk of muscular damage or inflammation (Cooper, 1983).

Subcutaneous injection can be difficult in birds, due to the thin skin that is quite inelastic, allowing fluids to leak through the puncture site (Coles, 1997). Several sites are available for subcutaneous injections in birds (see Fig. 9.4). The precrural fold or inguinal region is the site of choice for injection of larger volumes, for example fluid administration. Skin on the dorsal neck or interscapularly may also be used, particularly in larger birds. Other sites for small volumes are the flank, over the pectoral muscles, in the axilla, or in the propatagial skin fold of the wing (Harrison et al., 2006). Warm fluids will treat or prevent hypothermia and be less painful during administration than cold fluids. The addition of hyaluronidase at 150 units/L can be used to increase the absorption rate of subcutaneous medications (Chitty, 2005).

Venous access is useful for a number of reasons; phlebotomy may be performed, or fluids and drugs may be administered. The circulating blood volume in birds is between 6 ml and 12 ml per 100 g bodyweight (approximately 10%), and approximately 10% of this may be removed in a healthy bird without causing shock (Coles, 1997). For a 100 g cockatiel, the total blood volume would be approximately 10 ml, so 1 ml would be a maximum sample size; in practice, a smaller sample is taken, as most birds requiring phlebotomy are unwell. Birds appear to be more able to cope after blood loss than mammals (Kovách and Szász, 1968; Kovách et al., 1969).

Avian veins are fragile and easily traumatised, and haematomas rapidly form after venepuncture. Most veins are more easily accessed in anaesthetised birds, as manual restraint is stressful for the patient and allows movement that makes the procedure more technically difficult. If the patient is conscious during venepuncture, stress usually produces a higher blood pressure and there is an increased risk of haematoma following venous access; this often means that more prolonged restraint is necessary to apply haemostatic pressure following the procedure. The risks of anaesthesia, therefore, should be weighed against the benefits of venous access – be that to obtain a blood sample for investigative testing, to administer medication or fluids, or to place an intravenous catheter allowing more chronic venous access. Since birds may traumatise catheters, the catheter should be secured using sutures and/or tape and a light dressing.

The right jugular is the most easily accessible vein, and is useful for phlebotomy or emergency administration of drugs. The feathers are parted to expose the featherless aptera present in most species, and the skin prepared. Supporting the neck on the left-hand side helps stabilise the vein and tenses the skin overlying the vein. The vein is raised with gentle pressure at the base of the neck and the needle inserted at a shallow angle into the superficial vein (see Fig. 9.4). A 23–27-gauge needle is used, depending on patient size to reduce venous damage, with the bevel maintained uppermost during the procedure. Digital pressure should be maintained on the site, without occlusion of the trachea, for a few minutes after venepuncture to reduce the risk of haemorrhage or haematoma formation. Excess suction may collapse the vein and an appropriate size syringe should be used, for example a 0.5–1.0 ml syringe for passerines and 2.0–5.0 ml for psittacines.

The ulnar and brachial veins are subcutaneous at the elbow, on the ventral aspect of the wing (see Fig. 9.4). Access is easier in the anaesthetised patient, as the site can be more readily stabilised. Preparation of the skin with antiseptic in spirit is usually sufficient to allow visualisation of the vein. A 27- or 25-gauge needle or catheter can be used in most species to enter this vein. Intravenous catheters with wings for suture attachment are ideal, as the catheter can be secured to the skin or feathers; if the catheter does not have wings, adhesive tape can be used to manufacture a butterfly for attachment (see Fig. 11.3).

The superficial plantar metatarsal vein (see Figs 9.4 and 10.2) is useful for phlebotomy and catheterisation, particularly in birds with longer legs. The vein is raised by applied pressure over the medial distal tibiotarsus. Haematomas are less likely to form at this site after venepuncture due to the presence of scaly skin, but haemorrhage often necessitates temporary pressure, a dressing or tissue glue. This vein is more stable than the ulnar or brachial and may be catheterised in quite small birds.

Small blood samples may be obtained from toe-nail clipping, but urate contamination may affect chemistry results (O'Malley, 2005). Cardiac puncture is difficult in birds due to the protection of the keel, but can be performed in an emergency (Campbell, 1988).

Intraosseous injections enter the medullary cavity and thence access the vascular system, and are equivalent to intravenous administration. This route is extremely useful in small or collapsed birds where intravenous access is not possible. It is not advisable to use the intraosseous route in osteoporotic birds, as iatrogenic fractures may occur (Chitty, 2005).

In larger birds, a 20–22-gauge spinal needle may be used. Small intraosseous needles are available with insertion handles for ease of insertion. In smaller patients, a cheaper alternative is to use a hypodermic needle, although the lack of stylet may allow blockage with cortical bone during insertion. The distal ulna or proximal tibiotarsal bone is commonly used for intraosseous catheter placement (Figs 9.4, 9.11 and 9.12). The humerus cannot be used as it is pneumatised.

Usually the patient is anaesthetised, as insertion of a needle into bone is painful. The selected site is surgically prepared to reduce the risk of iatrogenic infection and resultant osteomyelitis. In patients with poor bone formation, iatrogenic fractures are also possible. The bone is

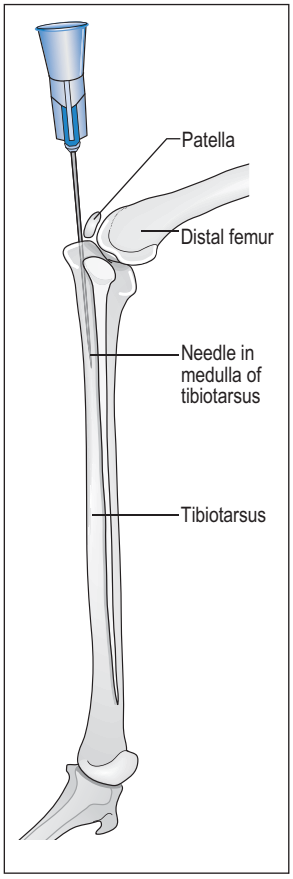


Figure 9.11 • Intraosseous needle placement in the tibiotarsus.

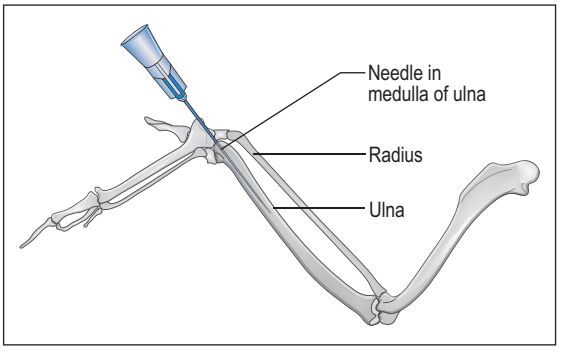


Figure 9.12 • Intraosseous needle placement in the distal ulna.

stabilised using the non-dominant hand and the needle gently inserted with a rotating movement into the bone.

To place a needle into the ulna, the carpus is flexed and the dorsal condyle proximal to the carpus is identified (see Fig. 9.12). The needle is inserted into the medullary cavity of the distal ulna in a proximal direction (Chitty, 2005). A fluid bolus injected into the ulna may be seen in the basilic vein.

To access the tibiotarsus, the stifle is flexed. The needle is inserted into the tibiotarsus from cranio-medially (Chitty, 2005). It is directed distally into the cranial cnemial crest, which lies distal to the stifle (see Fig. 9.11).

When the intraosseous needle is rotated, the bone surrounding the medulla can be felt with the needle tip. Injecting a small volume of sterile saline also assesses correct positioning; there should be no resistance to injection. Movement of the needle should result in similar movement of the bone; radiography will also show the needle position. The needle is secured using bandage material.

Fluids are injected intraosseously under pressure, either as bolus injections by hand or using a syringe pump. With regular flushing to maintain patency and appropriate sterility, intraosseous catheters can be used for up to 3 days (Chitty, 2005).

The *intraperitoneal* route is not commonly used in birds. True intraperitoneal injections should be administered in a paramedian location, with the skin and muscle raised using forceps. The needle is inserted at a very shallow angle to reduce the risk of perforating viscera. This route should not be used in conscious patients unless they are collapsed and unlikely to move during the injection (Coles, 1997). In general, fluids are not administered intraperitoneally in birds, due to the risk of drowning if an air sac is penetrated.

Other routes

Intratracheal administration of drugs may be appropriate, for example adrenaline (epinephrine) to treat cardiac arrest or saline for a tracheal wash. A small catheter, for example an intravenous catheter with the stylet removed, is used to introduce fluids into the trachea via the glottis. The maximum volume that should be administered via this route is 2 ml/kg body weight (Coles, 1997).

Topical medications are rarely used in avian species, as ointments and creams often contaminate the plumage and damage the feathers.

Many agents can be administered by *inhalation*. This includes volatile anaesthetics, but also therapeutic agents directed at the respiratory tract. Anaesthetics are usually administered via masks or endotracheal tubes; other drugs are often administered via nebulisation in a chamber. Droplet size must be below 5 μm in order to reach the air sacs (Coles, 1997).

It is less stressful in dyspnoeic patients to provide oxygen in a chamber rather than via a mask. For these patients, a (heated) humidifier should be incorporated (see Fig. 1.11) within the gas supply to reduce water loss from the airways and prevent drying of the respiratory tract (Benedikt et al., 1998). It is important to provide supplemental heating if a bird is in a chamber with gases flowing, as the patient may cool excessively.

Intubation

Placement of endotracheal tubes in birds is relatively easy. The glottis lies at the base of the tongue. The exact

location, therefore, varies depending on the anatomy of the tongue, with some species having a long thin tongue (see Fig. 11.4) and others having a short fleshy tongue (see Fig. 10.1). As described in the anatomy section, the lumen in avian tracheas is relatively large; for example, a 3–3.5 mm endotracheal tube may be used in a parrot weighing 300 g. As the tracheal cartilage rings are complete, uncuffed endotracheal tubes are usually used. The tube should be of an appropriate length such that when placed the connector lies either within or just outside the beak; this will minimise dead space within the circuit. It is often necessary to shorten the endotracheal tube before use in avian species.

The avian trachea has complete cartilaginous rings. Uncuffed endotracheal tubes are used in birds to reduce the risk of causing pressure necrosis.

Anaesthesia is induced using volatile or injectable agents. If a volatile agent has been administered via facemask, the mask is removed and the gases switched off during intubation. An assistant holds the upper and lower beaks open while the anaesthetist pulls the patient's tongue forward. The tongue may be grasped with your fingers, or atraumatic forceps, or dragged using a cotton bud or tongue depressor. A light source is used to visualise the glottis at the base of the tongue; a laryngoscope may be used in larger birds, or a pen torch or overhead light. It may help to apply pressure ventrally to raise the glottis from the base of the oral cavity. The endotracheal tube should be lubricated, for example with water-based KY Jelly® (Johnson & Johnson, New Jersey, US) or petroleum jelly, taking care not to block the lumen of the tube. The tube should be inserted when the glottis opens during a normal breath.

Endotracheal tube placement should be performed gently, to avoid injuring the delicate trachea. The endotracheal tube diameter should almost completely fill the lumen of the glottis and trachea, to reduce waste gases leaking into the environment. The tube size should enable a good seal with the glottis, but not be so tight as to cause damage. If the tube is loose in the trachea, a cuff may be inflated only with extreme care, as pressure necrosis is likely to cause stricture formation (Edling, 2006).

Until the endotracheal tube is secured to the patient, it should be held in place and the anaesthetic circuit reconnected to continue administration of anaesthetic gases. The tube is usually secured using adhesive tape, for example Micropore® (3M, Bracknell, UK). The tape is wrapped around the endotracheal tube within the oral cavity, then each end wrapped around the lower and/or upper beak (see Fig. 9.15). Alternatively, woven bandage can be used, attaching to the wings on the connector and then passing behind the bird's head to secure. This is preferable if the tube or patient is likely to become wet, which may affect the adhesive properties of tape.

Air sac cannulation

The air sac cannula bypasses the upper respiratory tract (see Fig. 9.7); for example, if an upper airway obstruction is present or if conventional endotracheal intubation would interfere with access for surgery on the head. Although anaesthesia is required for insertion of a cannula into an air sac (except in collapsed patients, when analgesia should still be administered), after insertion the cannula can then be used to provide oxygen or to maintain anaesthesia using volatile agents.

This is an invaluable technique for the avian clinician presented with a bird suffering from tracheal or syringeal obstruction. Anaesthesia via air sac perfusion has also been shown to be useful for ophthalmoscopic examination (Korbel et al., 1994). Most patients will not traumatise the cannula, particularly if the upper respiratory tract is obstructed (Edling, 2005). Air sac cannulation should not be performed in birds with air sacculitis or non-respiratory disease, such as ascites. This procedure is also not indicated in dyspnoeic birds with obstructions below the syrinx or lung disorders, such as polytetrafluoroethylene (PTFE) toxicity (Harrison et al., 2006). Care should be taken not to damage organs iatrogenically if organomegaly or gastrointestinal dilation is present.

Proprietary avian air sac surgical cannulae can be purchased (for example, 20 Fr non-cuffed tube with retention disc, Cook Veterinary Products, Bloomington, IN, USA), or shortened endotracheal tubes used for larger birds. In smaller species other tubing, such as urinary or intravenous catheters, can be shortened and used. Holes should be made in the side of the tube, to allow ease of ventilation and reduce the risk of obstruction. Ideally, the tubing should be sterile, but in an emergency cannulation with any piece of tubing will allow ventilation. The tube should be approximately the same diameter as the patient's trachea. The tube should be fairly short, to reduce the risk of iatrogenic damage to viscera during placement (Harrison et al., 2006).

The cannula is placed in the same site as that used for left lateral laparoscopy. The anaesthetised bird is placed in right lateral recumbency with the left leg pulled caudally, and a small area caudal to the last (eighth) rib just ventral to the vertebrae is surgically prepared (see Fig. 9.7A). In large birds, the cannula may be inserted between the last two ribs. The cannula is placed into the more superficial caudal thoracic air sac, rather than the deeper caudal abdominal air sac.

After preparation of the area, a small skin incision is made using scissors or an upturned scalpel blade (see Fig. 9.7B). Mosquito forceps are then used to dissect bluntly a small hole in the abdominal musculature into the abdominal cavity (see Fig. 9.7C). A 'pop' may be heard as the air sac membrane is penetrated underlying the muscular layer. By opening the forceps, the hole is enlarged sufficiently to allow insertion of the tube or cannula (see Fig. 9.7D). The tube is sutured to the skin to maintain its position. Proprietary avian air sac surgical cannulae have a retention disc that may be easily sutured to the patient's

skin. Correct positioning is ascertained by holding cotton wool or a down feather to the opening of the tube and checking for airflow with breaths (Coles, 1997; Harrison et al., 2006).

If anaesthesia is maintained via the air sac cannula, the normal airflow pattern may be altered on the left-hand side and the concentration of volatile anaesthetic agents often needs to be increased. If the interclavicular air sac is intubated, these airflow changes are not seen; however, it is more difficult to insert and maintain a cannula in this air sac. Total gas flow rates should be increased for air sac intubation, to ensure gases reach the lungs (Coles, 1997).

Assisted ventilation

Manual IPPV is used during most avian anaesthetics in order to maintain pulmonary perfusion and thence provide oxygen and volatile anaesthetic agents to the patient. If a mechanical ventilator is not available, compressing the reservoir bag within the anaesthetic circuit with the expiratory valve temporarily closed can be used to perform manual ventilation. Mechanical ventilators enable provision of anaesthetic gases at consistent pressures and frequency of respiration. Mechanical ventilation also frees the anaesthetist to monitor the patient more closely and provide other supportive care during the procedure.

Avian airway pressures should be less than 15–20 mmHg, or volutrauma of the air sacs will be produced (Edling, 2006). The pressure required to approximate normal inhalation will vary between individual animals. In most birds, 4–12 cm H_2O should be appropriate for tracheal or air-sac ventilation. Ideally, the ventilation rate should be adjusted to maintain $P_{ET}CO_2$ within normal limits, depending on minute-by-minute capnograph measurements. As a guide, adequate ventilation is usually present if the $P_{ET}CO_2$ is maintained between 30 and 45 mmHg (Edling et al., 2001).

In a study where chickens were anaesthetised with sevoflurane (Naganobu et al., 2003), controlled ventilation was found to prevent increases in $PaCO_2$ and heart rate that occurred during spontaneous ventilation. Another study in pigeons (*Columbia livia*) did not find clinically important cardiopulmonary changes when mechanical ventilation was used during routine anaesthesia, but respiratory alkalosis and cardiovascular depression occurred when air sac integrity was disrupted by coeliotomy (Touzot-Jourde et al., 2005).

Surgery on the coelomic cavity may involve penetration of air sacs. This will allow leakage of gases ventilated and will require adjustments on both pressure-limited and volume-limited mechanical ventilators. The respiratory system pressure will obviously be lowered if air sacs are open to the atmosphere. Volume-limited systems will not function well with a large leak such as this in the respiratory system, and will under-ventilate the patient. If mechanical ventilators are used during these procedures, the pressure or volume settings should be adjusted to increase gas flow in order to maintain sufficient anaesthetic gas within the respiratory tract.

Blood pressure measurement

The ulnar or distal ulnar (distal humerus) (see Fig. 10.3) or metatarsal (distal femur) arteries can be accessed to measure blood pressure indirectly in birds. The humeral site is more reliable in birds weighing less than 300 g. Small blood pressure cuffs are utilised, as the width should be approximately 40% of the circumference of the limb used. A Doppler probe is used to monitor blood flow audibly distal to the cuff. Birds with systolic blood pressures below 90 mmHg are treated for hypotension (with crystalloids and colloids) (Lichtenberger, 2005).

PRE-ANAESTHETICS

Pre-medications are rarely administered to birds, but some agents may assist smooth induction and recovery from anaesthesia, have volatile agent-sparing effects, and/or provide analgesia.

Opioid administration will reduce the concentration of volatile agent required to induce and maintain anaesthesia. Individual species variation in opioid receptors will affect the response seen with opioid agents; for example, most opioid receptors in pigeons are of the κ type and butorphanol may be a better analgesic in this species than μ opioid agonists such as morphine (Mansour et al., 1988). Analgesics, such as butorphanol, will also produce some sedative effects.

Studies in dogs undergoing surgery have shown that hydromorphone administration may produce a transient increase in $PaCO_2$ postoperatively, and that administration of hydromorphone or butorphanol may result in transient reduction in PaO_2 (Campbell et al., 2003). While these changes appear to be mild in mammalian species, similar physiological changes associated with opioid use within avian species may be significant (Edling, 2006).

The *benzodiazepines* midazolam or diazepam may be used before anaesthetic induction with other agents. These tranquillisers produce excellent muscle relaxation, but do not provide analgesia. Midazolam may be administered intramuscularly to tranquillise a bird and reduce the stress of induction with a volatile agent. This will decrease the levels of circulating catecholamines, reducing the risk of catecholamine-induced cardiac dysrrhythmias (Edling, 2005). Unlike midazolam that is absorbed rapidly and almost completely from the muscles, uptake of diazepam from the intramuscular site is slow and unpredictable. Diazepam may be administered intravenously to produce the same effect (Ludders and Mathews, 1996). Midazolam is more potent and longer-lasting than diazepam and does not have adverse effects on mean arterial blood pressure and blood gases (Edling, 2006).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

General approach to avian anaesthesia

Larger birds should be fasted (see Peri-anaesthetic section) before anaesthesia to reduce the risk of regurgitation. If emergency anaesthesia is required in an unfasted bird, the trachea should be intubated, particularly if the crop is full or the bird has recently been fed. Small birds are not normally fasted, due to the risks of hypoglycaemia. On induction, the oropharynx should be checked for the presence of food or other foreign material that could be inhaled and the crop palpated to check for ingesta.

Oxygen should be supplied to all anaesthetised patients. For brief procedures lasting less than 10 min, a facemask is sufficient, although an appropriately sized endotracheal tube should be on hand in case it is required. The facemask should be closely fitting around the bird's neck to allow PPV and to reduce environmental contamination of waste gases. Patients should be intubated for most procedures to maintain a patent airway, for administration of oxygen and volatile agents, and to permit PPV. If non-elective anaesthesia is indicated in a bird with a full crop, the crop contents should be aspirated before induction if possible and intubation performed to protect the airway from aspiration of regurgitated ingesta.

In patients with air sac cannulation, the anaesthetic circuit, for example an Ayre's T-piece (see Fig. 9.7E), can be attached directly to the cannula as it would be for an endotracheal tube. The gas flow rates are reduced to one-third that used during anaesthesia via tracheal intubation. PPV can be used, including mechanical ventilators. However, normal respiratory movements usually stop and anaesthetic monitoring is more difficult in these patients (Edling, 2005).

Induction

Most birds are induced with volatile anaesthetic agents, using a facemask or induction chamber. The choice of volatile agent may depend on those available in the veterinary practice, but isoflurane or sevoflurane is commonly used. It may be appropriate to pre-medicate the bird beforehand, particularly if a prolonged procedure is planned. Injectable agents are discussed as they may be useful adjuncts to volatile agents in certain cases, but are rarely used as sole agents for avian anaesthesia.

Injectable agents

Injectable anaesthetic agents are uncommonly used to induce and maintain anaesthesia in birds. There are several problems associated with injectable anaesthetics, not least of which is the risk of overdosage. An accurate weight should be obtained prior to the use of any injectable agents in birds. The presence of plumage variably covering the body makes weight estimation very difficult in birds, and overdosage is exceptionally easy in small birds. Small volumes of drug are difficult to measure; dilution of the agent with sterile water for injection or saline and the use of insulin syringes will improve the accuracy of drug measurement.

Other problems associated with the use of injectable anaesthetic agents in birds include delay in onset of anaesthesia, species and individual variability in response to agents, cardio-respiratory depression, slow induction and prolonged and/or traumatic recoveries (Ludders and Mathews, 1996). Injectable agents are thus rarely used in veterinary practices, as inhalational agents are much more reliable and safer to use.

Injectables may be used in conjunction with volatile anaesthetics, or where sedation or anaesthesia is required outwith the practice premises where equipment is stationed. Injectable drugs may be cheaper than volatile agents. Patients should ideally still be intubated after induction with injectable anaesthetics and oxygen provided if possible. Monitoring is performed similarly for injectable and inhalation anaesthesia. Some agents are used in combination protocols to reduce the concentrations of volatile agent required, for example analgesic agents that also produce sedation.

Although *opioids* are predominantly used to provide analgesia for patients undergoing surgery or with painful conditions, they may be used to augment the anaesthetic protocol.

Alfaxalone/alphadolone can be administered by several routes, including intravenously, intramuscularly and intraperitoneally. This drug has been used in many species. Intravenous administration produces the most predictable anaesthesia, with good muscle relaxation (Coles, 1997; Cribb and Haigh, 1977). However, cardiac abnormalities have been recorded – some were fatal (Cooper and Redig, 1975).

Propofol has been used in many species (Fitzgerald and Cooper, 1990). It is non-irritant to tissues and is very rapidly metabolised, but has a narrow safety margin (Coles, 1997). Respiratory depression and apnoea may result from an overdose, and supplemental oxygenation and PPV are advisable (Machin and Caulkett, 2000). Propofol may be used in larger birds, with or without pre-medication, to induce anaesthesia and allow intubation prior to maintenance on volatile anaesthetic agents. Administration is by slow intravenous injection, producing rapid onset of anaesthesia. Up to 10 mg/kg is required to induce anaesthesia and incremental doses of up to 3 mg/kg may be used to prolong anaesthesia (Edling, 2005). The lack of residual and cumulative effects provides rapid recovery and allows supplemental doses to be administered to prolong anaesthesia. As in other species, dose-dependent cardio-respiratory depression may be seen.

Ketamine is a useful agent in birds. It can be administered via any parenteral route, and is often used in combination with other drugs. This agent will produce chemical restraint and moderate analgesia, but is unsuitable for major surgical procedures when used alone (Ludders et al.,

1989b). Ketamine induces anaesthesia of 10–30 min duration 3–5 min after intramuscular administration. Recovery is variable, from 30 min to 5 h. The dose varies greatly between species, with higher doses required in smaller birds. Increasing the dose of ketamine administered does not increase the depth of anaesthesia, but prolongs the effects (Edling, 2005).

Ketamine is usually administered intramuscularly, with anaesthesia occurring within 3–5 min and persisting for approximately 35 min. Initially inco-ordination, opisthotony and relaxation are seen (Coles, 1997). Ketamine may also be administered intravenously to induce anaesthesia in larger birds, usually in combination with other agents. Administration should be slow and incremental, as a rapid overdose may result in apnoea and/or cardiac arrest. When used in combination with other agents, such as xylazine, repeating the ketamine dose will extend the period of anaesthesia. Once more, this will prolong the recovery period (Edling, 2005).

The main disadvantage associated with ketamine is cardio-respiratory depression, which increases with higher doses (Coles, 1997; Edling, 2005). There are significant variations in the response to ketamine between avian species (Ludders et al., 1989b). Thermoregulation may be affected (Altman, 1980). Hepatic metabolism and renal excretion of this drug mean that it should be avoided in birds with hepatic or renal dysfunction, or where dehydration is suspected.

Ketamine has been used in birds with *acepromazine*, but both drugs induce bradycardia (Coles, 1997).

The benzodiazepines, *midazolam* or *diazepam*, may be used as lone agents or in conjunction with ketamine. The benzodiazepine–ketamine combination is administered intramuscularly and produces deep sedation or anaesthesia with good muscle relaxation. The addition of a benzodiazepine to the ketamine also smoothes induction and recovery (Edling, 2005). Respiratory depression is seen with these agents (Coles, 1997).

Alpha-2-adrenergic agents, such as *xylazine* and *medetomidine*, are not usually used as sole agents, although they have sedative and analgesic properties. Side effects are species-dependent. Xylazine use may be associated with respiratory depression, hypoxaemia, hypercapnia, excitement or convulsions (Ludders and Mathews, 1996; Ludders et al., 1989b). Cardio-respiratory effects with the alpha-2-agonists may be severe, including second-degree heart block, bradyarrhythmias and sensitisation to catecholamine-induced cardiac arrhythmias (Coles, 1997; Edling, 2006).

Ketamine enhances the sedative and analgesic effects of xylazine, producing anaesthesia in many species, along with reasonable muscle relaxation and only slight respiratory depression. The addition of ketamine enhances the sedative and analgesic effects of xylazine (Edling, 2006). With this combination, the palpebral reflex may be absent or slow. Increasing the ketamine dose will increase the depth and duration of anaesthesia, but also prolong the recovery time (Coles, 1997). The drugs may be given intramuscularly or intravenously, or variations of both.

Some studies found cardiac and respiratory disturbances when these were administered intravenously, and it is recommended to use the intramuscular route (Haigh, 1980; Redig, 1983). Coles reports using a dose of 20 mg/kg of ketamine and 4 mg/kg of xylazine, administered intramuscularly, in several species (Coles, 1997). This produces sedation within a few minutes, induction in 5–7 min, anaesthesia for 10–20 min and recovery to perching in 1–2 h. Larger species appear to require a lower dose of ketamine.

There may be incoordination and excitement during recovery, and it is advisable to wrap the bird loosely at this stage. This combination is, therefore, unsafe in certain species, such as long-legged species that may damage themselves during this period.

Ketamine may be given intramuscularly with medetomidine to induce anaesthesia in several species (Jalanka, 1991; Reither, 1993; Scrollavezza et al., 1995). As with xylazine, medetomidine causes sedation, muscle relaxation and analgesia. Side effects include bradycardia and peripheral vasoconstriction. Medetomidine is more potent than xylazine and has a wide safety margin. Induction of anaesthesia occurs in 2–3 min and anaesthesia can be maintained with low doses of volatile anaesthetic agents (Coles, 1997). Surgical anaesthesia induced in pigeons with ketamine, medetomidine and butorphanol produced arrhythmias in some birds, which ceased after atipamezole administration (Atalan et al., 2002).

Atipamezole and *yohimbine* are alpha-adrenergic antagonists and are used to reverse medetomidine and xylazine, reducing the recovery time significantly or for use in an emergency to treat an overdose. If a combination containing ketamine has been administered, the alpha-2-antagonist should not be administered until 30–40 min has passed. Administration of the reversal too early may result in violent wing flapping due to the residual effects of ketamine and re-sedation may occur, as atipamezole is more rapidly metabolised than medetomidine. It is usual to administer the same dose of atipamezole as medetomidine, but higher doses may reduce the risk of re-sedation (Coles, 1997).

Neuromuscular blockers are not routinely used in avian patients, but *vecuronium* at 0.2 mg/kg has been shown to optimise mydriasis for ophthalmoscopic examination (Korbel et al., 1997). Used concurrently with isoflurane anaesthesia, the vecuronium enabled the isoflurane concentration to be reduced by 25% (Korbel, 2000).

Volatile agents

Volatile agents are the most common form of anaesthetic used in avian species. Their many benefits include rapid induction and recovery with few adverse side effects at doses producing a surgical plane of anaesthesia (Edling, 2005). Anaesthesia can be controlled easily using modern vaporiser units and anaesthetic circuits. Most species are readily intubated to facilitate provision of anaesthetic gases; this also permits PPV to be performed. The main side effects seen with volatile anaesthetic agents are dose-dependent depression of the central nervous, cardiovascular and respiratory systems.

As birds do not have an alveolar lung, the term 'minimum alveolar concentration' (MAC) is not appropriate. MAC in birds is, therefore, defined as the minimum

Table 9.5: Injectable anaesthetic agents in birds

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	COMMENT
Alphaxolone/alphadolone	Raptors	10[4]	IV	Cardiac irregularities reported
Atipamezole	All	<5 × alpha-2-agonist dose[9]	IM	Reversal of alpha-2-agonists (medetomidine or xylazine)
Atropine	Most species	0.02–0.08[6] 0.01–0.02[6]	IM IV	Parasympatholytic agent
Butorphanol	All	0.51.0[1]	IM	Sedative pre-medication at lower dose May cause respiratory depression
Butorphanol + ketamine + medetomidine	Psittacines	1 + 3 + 0.04[13]	IM	Pre-medication, reduces volatile agent requirements and improves ventilation
Diazepam	Most species	0.2–0.5[1]	IM, IV	Aqueous solution IM or IV; propylene glycol solution IV only Tranquillisation, pre-medication
Glycopyrrolate	Most species	0.01–0.02[6]	IM, IV	Parasympatholytic agent
Ketamine	All Budgerigar Raptor	50–100[10] 50[3] 2.5–170[3]	IM	Used on 3 consecutive days without side effects Induction of anaesthesia Poor muscle relaxation and violent recoveries, so rarely used alone
Ketamine + diazepam	Most species	5–30 + 0.5–2.0[2]	IM + IM, IV	Anaesthetic induction
Ketamine + medetomidine [3]	Diurnal raptors Owls Psittacines	5 + 0.1 10 + 0.1–0.15 5 + 0.075	IM	Deep sedation/anaesthesia
Ketamine + midazolam	Most species	10–40 + 0.2–2.0[10]	SC, IM	Anaesthesia
Ketamine + xylazine	Most species	2.5–5.0 + 0.25–0.5[7]	IM	Cardiac depression, rough recovery. Much species variation. Higher doses of ketamine for smaller birds
Midazolam[1]	Most species	0.1–0.5 0.05–0.15	IM IV	Tranquillisation
Propofol	All (variable dose required across species)	<14[5]	IV	Induction, dose to effect. Respiratory depression, so intubate and PPV Incremental bolus to prolong anaesthesia
Tiletamine/zolazepam	Most species	10–30[8]	IM	Restraint, anaesthesia Prolonged, rough recoveries

(*Continued*)

Table 9.5: (Continued)

DRUG	SPECIES	DOSE (MG/KG)	ROUTE	COMMENT
Xylazine	Raptors, psittacines	1.0–2.2[11]	IM, IV	Heavy sedation Rarely used due to common side effects
Yohimbine	Most species	1.0[12]	IV	Reversal of alpha-2-agonists (medetomidine or xylazine) Excitement and mortality at higher doses

Key: IM = intramuscular, IV = intravenous, PPV = positive pressure ventilation, SC = subcutaneous
[1] (Abou-Madi, 2001); [2] (Bishop, 2001); [3] (Coles, 1997); [4] (Cooper, 1978); [5] (Fitzgerald and Cooper, 1990; Huckabee, 2000); [6] (Edling, 2005); [7] (Haigh, 1980); [8] (Hartup and Miller, 1992); [9] (Jalanka, 1991); [10] (Mandelker, 1972, 1973); [11] (Mandelker, 1988; Marx and Roston, 1996); [12] (Samour, 2000); [13] (Teare, 1987)

anaesthetic concentration required to keep a bird from purposeful movement to a painful stimulus (Ludders et al., 1989a). MAC values will vary slightly between bird species, for example the MAC for isoflurane is 1.44% in cockatoos and 1.32% in ducks (Ludders and Mathews, 1996; Ludders et al., 1990). Values from studies in various species are listed below as a guide.

An alternative comparison of anaesthetic agents is the Anaesthesia Index (AI), which compares agents' abilities to produce respiratory depression and apnoea. For this index, a lower value corresponds to a higher risk of apnoea (Edling, 2006). Comparison of this index in various species has shown birds to be much more sensitive to the respiratory depressive effects of isoflurane than mammalian species, with an AI of 1.0 in ducks (Ludders et al., 1990) compared to 2.51 in dogs (Steffey, 1978). This emphasises the need to monitor anaesthesia closely in birds. Respiratory depression occurs with all agents with the concentrations required to produce a surgical plane of anaesthesia (Ludders et al., 1989a).

Gaseous agents are routinely used to produce anaesthesia in avian species. They allow rapid alterations of anaesthetic depth in response to varying stimuli or patient response, allow rapid induction and recovery, and have few side effects (Muir and Hubbell, 2000a). Isoflurane and sevoflurane are the most commonly used volatile anaesthetic agents in birds. Isoflurane, sevoflurane and desflurane all produce dose-dependent effects on the central nervous, respiratory and cardiovascular systems (Edling, 2006).

With larger birds, it is usual to restrain the patient in a towel and induce by administering a volatile anaesthetic agent via a facemask. The efficient avian respiratory system means that anaesthetic induction by this technique is rapid. This enables the clinician to assess continuously the patient's respiratory movements, heart rate and reflexes. When applying the mask, avoid damaging the patient's eyes and beak. The mask should surround the bird's head, with the rubber ring on proprietary masks snug (but not excessively tight) around the neck. Unfortunately, many facemasks do not create a tight seal with the avian head, and the risk of environmental contamination with waste gases may be considerable. Cotton wool or swabs may be used to try to reduce environmental contamination, but most patients are intubated for maintenance after induction using a facemask.

For small birds, an anaesthetic chamber may be used for induction. This is often less stressful than restraining the patient for mask induction. The chamber may be purpose-built, or more usefully an adapted plastic container. Plastic drinks bottles are easily adapted (see Fig. 1.5) and various sizes can be made to accommodate a range of patient sizes. The use of a smaller chamber allows the gas concentration to be varied more readily, small birds will be less able to traumatise themselves during induction than in a large chamber and there will be less waste gas released into the environment when the bird is removed from the chamber. The main disadvantage with chamber induction is the difficulty in monitoring the depth of anaesthesia during induction, as the anaesthetist cannot easily check muscle relaxation or auscultate the cardio-respiratory system. As the patient is not restrained, it may injure itself during the excitement phase of induction (Edling, 2005). A clear chamber is essential, so the patient can be observed to monitor when the righting reflex is lost, checked by gently rolling the chamber.

It is normal initially to preoxygenate the patient prior to induction using either of the above techniques. After a few breaths in 100% oxygen, the anaesthetic agent is switched to a high concentration (Table 9.6). This usually results in rapid induction of anaesthesia with minimal stress for the patient (Edling, 2006). If a gradual increase in concentration of anaesthetic agent is used, induction is slower and the patient may become stressed due to the prolonged restraint, with concomitant catecholamine increases (Edling, 2005). Once anaesthesia is attained, the concentration of volatile agent is reduced to a level at or just below its MAC for maintenance.

Volatile anaesthetic agents may depress the $PaCO_2$ chemoreceptors, reducing the respiratory rate in the anaesthetised patient (Coles, 1997). Ventilation is, therefore, usually assisted either manually or using a mechanical ventilator.

Table 9.6: Inhalation anaesthetics in birds

DRUG	INDUCTION CONCENTRATION*	MAINTENANCE CONCENTRATION (%)	COMMENT
Desflurane	6–8[2]	4–6	Odour precludes ability to induce via facemask
Halothane	0.5–3.0 (incremental)	2–3	Induction in 2–5 min
Isoflurane	4–5[1,2,3]	2.5–3.0	Induction in 2–3 min
Nitrous oxide	50:50 in oxygen[1]	Maximum 50:50 in oxygen	Discontinue 5–10 min before end of anaesthesia
Sevoflurane	7–8[2,3]	4[4]	Smooth induction as no respiratory tract irritation

* Lower concentrations should be used in compromised patients
[1] (Coles, 1997); [2] (Edling, 2006); [3] (Edling, 2005); [4] (Helmer, 2004)

Halothane has been used historically in many species without problems (Graham-Jones, 1966; Jones, 1966; Marley and Payne, 1964). However, there are reports of overdosage with this agent (Jones, 1977). Side effects include direct myocardial depression, causing sensitisation to catecholamine-induced cardiac arrhythmias. These arrhythmias may be fatal, particularly in stressed individuals with high levels of circulating catecholamines (Edling, 2006). One study in ducks anaesthetised with halothane suggested that the arrhythmogenic effects seen were related to hypercapnia (Naganobu et al., 2001). The drop in body temperature in birds anaesthetised with halothane appears to be more significant than patients where other agents are used. A major problem with halothane is that cardiac and respiratory arrest are simultaneous with this agent (Coles, 1997).

If halothane is utilised, it is safer to increase the concentration of halothane gradually during induction, up to 3%. If 3–4% is used initially, apnoea may occur with a high concentration of anaesthetic agent in the posterior air sacs (Coles, 1997), with an associated risk of overdose. Halothane undergoes partial (20%) hepatic metabolism, and is not recommended in patients with suspected hepatic dysfunction.

Isoflurane is considered a safe anaesthetic in avian species and is currently the most commonly used agent in veterinary practices. The MAC value for isoflurane in cockatoos is 1.4 (Ludders and Mathews, 1996). The blood gas partition coefficient is 1.41 (Edling, 2006), and blood–brain tissue coefficient 1.7 (Franchetti and Kilde, 1978). Induction and recovery are rapid due to this relative insolubility in plasma; alterations in the anaesthetic depth are also rapid (Abou-Madi, 2001).

Isoflurane has several advantages over halothane. A significant benefit is that cardiac arrest does not occur for several minutes after respiratory arrest with isoflurane, allowing the anaesthetist to intervene. Myocardial effects are also much less than those seen with halothane, as isoflurane does not sensitise the myocardium to catecholamine-induced arrhythmias (Muir and Hubbell, 2000b). Only 0.3% of isoflurane is metabolised in the liver; therefore, it is much safer for patients with hepatic dysfunction and for veterinary staff who may be exposed to leaked gases (Coles, 1997).

A number of side effects may be seen with isoflurane. Some respiratory tract irritation occurs, which may result in stress during induction (Edling, 2006). A decrease in peripheral vascular resistance results in a lowered blood pressure (Coles, 1997). Doppler-derived blood flow measurements in common buzzards (*Buteo buteo*) have shown isoflurane anaesthesia to reduce blood flow velocity as well as heart rate (Straub et al., 2003a); these changes are likely also to be caused by other anaesthetic agents. Patient age and gender may affect arterial pressures in certain species (Ruiz-Feria et al., 2004). Isoflurane has been reported to cause arrhythmias, for example second- and third-degree atrioventricular block, sinus arrest, T-wave depression and atrial premature contraction (Aguilar et al., 1995).

Respiratory depression may still be seen with isoflurane, in part due to the muscle relaxation caused by this agent (Straub et al., 2003a). $PaCO_2$ increases with increasing isoflurane concentrations, producing a respiratory acidosis (Ludders et al., 1989a). As with other agents, PPV is, therefore, advisable to prevent hypercapnia (Forbes, 1999).

Induction with isoflurane occurs in 1–2 min when a concentration of 3–5% is administered via a close-fitting facemask. Recovery is similarly rapid, although it appears to be related to the period of anaesthesia, being slower in patients that have been anaesthetised for a long period of time (Edling, 2005).

Sevoflurane has a lower blood gas partition coefficient (0.69) than isoflurane, resulting in lower blood solubility (Edling, 2006). Sevoflurane is less potent than isoflurane, with an MAC for sevoflurane in chickens of 2–3%

(Naganobu et al., 2000). The blood–brain tissue coefficient is 1.7 (Edling, 2005). Lower solubility and MAC result in shorter recovery time compared to isoflurane (Greenacre and Quandt, 1997). If a critical patient requires anaesthesia or a prolonged anaesthetic is envisaged, sevoflurane is considered a better option than other agents, as the recovery will be faster. The time to perching is quicker and the birds are less ataxic after sevoflurane anaesthesia compared with isoflurane.

Sevoflurane does not cause respiratory tract irritation and mask induction is, therefore, less stressful than isoflurane and desflurane (Edling, 2006). A disadvantage is the higher current cost of sevoflurane over other agents. Sevoflurane also depresses plasma ionised calcium levels (Edling, 2005; Stanford, 2003). A decrease in arterial blood pressure was observed during one study using sevoflurane anaesthesia in chickens, which was more significant during controlled than spontaneous ventilation, probably due to hypercapnia occurring during spontaneous ventilation (Naganobu et al., 2000; Naganobu et al., 2003).

Desflurane has very low blood solubility, with a blood gas partition coefficient of 0.42, but it is less potent than the other agents, with a MAC of 6–8% (Edling, 2006). Tissue solubility is also low in desflurane, with a blood–brain tissue coefficient of 1.3 (Franchetti and Kilde, 1978). These values produce an agent with a faster recovery time than the other volatile agents, particularly after prolonged anaesthesia (Edling, 2006). The main problem with desflurane is the pungent odour that precludes mask induction and this may limit its usefulness in avian anaesthesia. A specialised temperature-controlled pressurised vaporiser is also required to use this agent. As with isoflurane, some respiratory tract irritation occurs with desflurane, which may result in some stress during induction. Due to these issues, desflurane is not used for mask induction (Eger, 1993).

Nitrous oxide may be used to reduce the concentrations of other volatile agents required for anaesthesia, using a maximum of 50% nitrous oxide (Korbel et al., 1994; Korbel et al., 1997). It is not sufficiently potent to produce anaesthesia when used as the sole anaesthetic agent (Edling, 2005). This agent depresses ventilation at surgical anaesthetic levels (Ludders et al., 1989a), increasing $PaCO_2$ and producing respiratory acidosis (Scheid and Piiper, 1989). Nitrous oxide should be ceased 5–10 min before the end of anaesthesia. Oxygen should be the sole carrier gas in patients with respiratory disease (Coles, 1997).

Anaesthetic maintenance

After induction, most avian species can be intubated. Intubation allows the clinician to have more control over the bird's respiratory system, with airway patency assured and allowing PPV to be peformed. There is also less risk of waste gas escape into the environment compared to when using facemasks.

Intubation may not be performed if a short (less than 10 min) non-invasive procedure is planned; for other procedures it is a valuable technique. As discussed above, avian anatomy allows the clinician to provide anaesthetic gases either via the trachea or from the air sacs. Cannulation of the air sacs will affect the normal respiratory physiology, and a minor increase in anaesthetic gas concentration may be required to maintain anaesthesia. However, the direction of airflow does not adversely affect gas exchange (Ludders et al., 1989a). Similarly, if endoscopy is being performed via a caudal air sac while the patient is anaesthetised using an endotracheal tube, the anaesthetic gas flow rate and concentration should be increased slightly to account for gas leakage via the endoscopic access site.

During anaesthesia, administration of volatile agents will rapidly deepen the level of anaesthesia. If the plane of anaesthesia is too deep, lower the concentration of volatile agent and consider reversal of injectable agents.

Recovery

Volatile anaesthetic agents are switched off, and the patient is disconnected from the anaesthetic circuit while it is flushed with fresh oxygen. The circuit is then reconnected to the bird to provide 100% oxygen during recovery (Edling, 2006). The tape or bandage holding the endotracheal tube in place is loosened to allow for rapid removal of the tube; the anaesthetist should hold the tube in place at this point.

If volatile agents have been used alone for anaesthesia, recovery should be rapid. If injectable anaesthetic agents have been administered, reversal agents should be given. As the plane of anaesthesia lightens, the bird will have muscle fasciculations; this may reduce the audibility of heart sounds (Edling et al., 2001). Muscle tension then returns with spontaneous wing and leg movements. As jaw tone returns along with coughing and swallowing, the endotracheal tube should be removed to prevent the bird from biting through it.

If possible, the bird should be restrained in a normal position during the recovery phase, using a towel to support and restrain the bird gently in an upright position. Excess restraint will prohibit breathing movement and may also lead to hyperthermia. Oxygen should be provided via an open mask with the rubber diaphragm removed held to the head after extubation until the bird can hold itself upright. If anaesthesia has been brief, the patient may be able to return directly to its original cage or carrier, but a towel should be placed on the floor and high perches removed until the patient has recovered sufficiently to perch steadily.

During recovery, the bird's environment should be kept quiet and slightly darkened to reduce stressful stimuli. The bird should be fully recovered from anaesthesia within a few minutes to 2 h, depending on the length of anaesthetic (Edling, 2006). This will depend on the patient's condition, the length of anaesthetic, the anaesthetic agents used and the procedure performed. If recovery is not rapid, review the patient's status and provide supportive treatment as required.

Supplemental heating should be provided until the patient is moving around normally. A pre-warmed

incubator or brooder with oxygen supplementation (see Fig. 9.5) and a padded floor are ideal for recovery after a long procedure, as recovery may be more prolonged.

Suggested anaesthetic protocols

The anaesthetic protocol selected will depend on the reasons for anaesthesia. If a painful procedure is to be performed, analgesia should be provided. Hypnotic agents are useful to sedate patients, but are not good muscle relaxants and, therefore, not so useful during surgery. As with other groups of animals, there is much interspecies variation in response to anaesthetic agents. Individual variation will also affect how the patient responds to various agents, likely due to variations in hepatic and plasma enzymes affecting detoxication and excretion (Coles, 1997). Pre-existing hepatic and renal disease will also affect drug metabolism.

Anaesthesia for short procedures in birds is usually achieved using volatile anaesthetic agents. The most common drug used is *isoflurane*, as induction and recovery are rapid, and most of the agent is excreted unmetabolised via the respiratory tract. In most species, a facemask can be used to deliver the anaesthetic gas, but a chamber is used to induce anaesthesia in very small birds that may be stressed by the restraint necessary for mask induction. It is useful to pre-oxygenate patients with 100% oxygen for a few breaths, to counteract the transient apnoea that may occur during induction. If isoflurane is used, the concentration of inspired anaesthetic agent is then turned on at 3–5%. Most patients will be induced within 2 min at 3% isoflurane, but a few require higher concentrations. Lower concentrations of volatile anaesthetic should be used in compromised patients, as they will be more susceptible to adverse effects. If sevoflurane is used, 4–5% should be administered to ill patients rather than the 8% used in 'healthy' birds (Helmer, 2004).

Anaesthesia may be maintained via a facemask at 2–3%. This is suitable if a very short procedure, for example phlebotomy, is to be performed. The bird should be intubated if it has recently eaten or a more prolonged procedure is to be performed, for example radiography. IPPV should be performed to ensure adequate ventilation.

It may be appropriate to use *injectable agents* in conjunction with volatile anaesthetics if a more prolonged anaesthesia is anticipated, for example orthopaedic surgery. This has the advantage of providing a more balanced anaesthetic, lowering the concentration of volatile agents required, and enabling the clinician to consider the addition of an analgesic to the protocol if required. Injectable agents may be used as pre-medicants, for example midazolam and butorphanol. Alternatively, injectables may be used to induce anaesthesia, for example ketamine with medetomidine or diazepam (Coles, 1997). If combinations are administered, the doses of individual drugs required to produce anaesthesia will be reduced.

After induction with injectable agents, anaesthesia is 'topped up' or maintained using volatile agents via mask or endotracheal tube. IPPV is again used to maintain respiration that is depressed during anaesthesia. As respiratory depression is common when using injectable agents, oxygen should always be supplemented when using these drugs.

Anaesthesia in neonatal or paediatric patients

These patients require extra care when anaesthetised. Although induction and recovery are usually rapid, careful monitoring is required to detect and correct problems swiftly.

The glottis and trachea are soft and easily damaged, and intubation and extubation should be performed carefully. Oxygen requirements are higher in these birds. Gas flow rates should, therefore, be two to three times higher than rates provided for adults to prevent hypoxia and avoid carbon dioxide build-up.

Cardiovascular effects during anaesthesia are more serious in younger birds than adults and careful monitoring is essential to detect problems early. Haemorrhage may result in tachycardia and hypotension.

There is a high risk of hypoglycaemia in young patients. These birds should not be fasted, but the crop should be emptied by aspiration just before induction. If anaesthesia is prolonged or the patient is not self-feeding soon after the procedure, crop feeding should be performed to prevent hypoglycaemia. A maximum of 2 h should pass between feeds. Featherless chicks are prone to hypothermia and require supplemental heating (Forbes and Altman, 1998).

ANAESTHESIA MONITORING

Due to the rapidity of change in anaesthetic depth with changing anaesthetic concentration or response to stimulation in birds, it is imperative to monitor the avian patient closely throughout the anaesthetic period. The parameters listed in the sections below will allow the anaesthetist to monitor the patient's progress under anaesthesia. The use of equipment such as electrocardiography, capnography and Doppler flow monitoring improves patient monitoring. As in other species, deteriorating trends in parameters indicate a deterioration of physiological state. In birds this deterioration can rapidly be fatal and interventions must be performed without delay.

Observations on the patient

Cardiovascular system

The heart rate and rhythm should be monitored, using a bell or (in larger patients) oesophageal *stethoscope*. In larger patients, a peripheral *pulse* can be palpated or monitored using a Doppler flow probe. Suitable pulses palpable in most birds are the brachial artery in the axillary region or the dorsal metatarsal artery over the tarsometatarsal bone.

As respiratory movements usually cease during air sac cannulation, anaesthesia monitoring in these patients is

predominantly based on cardiovascular assessment. Heart rate and peripheral blood flow should be assessed (Edling, 2005). Capillary refill time should be less than 1 s. It can be assessed on non-pigmented skin on the feet, or on the basilic or ulnar veins at the medial elbow. Mucous membrane colour can be monitored in anaesthetised birds, but cannot be relied upon to assess peripheral oxygenation in critical patients.

Respiratory system

Ventilation can be assessed by monitoring respiratory rate, depth and rhythm. These can be measured by directly observing the patient or movements of the anaesthetic reservoir bag. A bell stethoscope can be used to auscultate movements associated with respiration within the coelomic cavity, or an oesophageal stethoscope can be placed in larger birds. The respiratory rate is likely to slow during anaesthesia, but should not be less than half the conscious resting rate (Coles, 1997). It is not possible to ascertain adequate ventilation via respiratory rate and tidal volume, and equipment (described below) should be used in conjunction with these techniques.

Respiratory rate and depth are also useful measures of anaesthetic depth (Edling, 2006), but may be difficult to assess if PPV is being performed. The respiratory pattern will be rapid and irregular at light planes of anaesthesia, becoming slow and regular during surgical anaesthesia.

The plane of anaesthesia is too deep if respiration becomes rapid, shallow or intermittent (Coles, 1997; Forbes and Altman, 1998). Respiratory depression, associated with anaesthetic agents or other drugs, such as opioids, may cause hypercapnia. Hypoxia affects different species to different degrees; for example, birds that normally fly high will be more adapted to hypoxia than lower-ranging species (Dawson, 1975). Respiratory depression by anaesthetic drugs may be more concerning in small passerines than larger species.

Nervous system

Various reflexes can be assessed to ascertain the plane or depth of anaesthesia. The depth of anaesthesia required will depend on the procedure being performed. Obviously a light plane will be sufficient for restraint to take a blood sample, but a deeper plane is necessary to perform surgery. As with all anaesthetics, the anaesthetist should monitor reflexes closely, as their absence may indicate a life-threatening situation.

Reflexes are similar to other species, and include eye reflexes, jaw tone, the pedal reflex, cloacal reflex and muscle relaxation. There will be individual bird variation to stimulation of reflexes (Coles, 1997).

The first reflex to be lost on anaesthetic induction is the righting reflex. At a light-to-medium plane of anaesthesia, the response to stimulation of the cere and peri-cloacal skin will just be abolished. The corneal reflex is a reliable indicator of anaesthetic depth in birds and may become sluggish, but should never be lost during anaesthesia (Forbes and Altman, 1998). Stimulating the interdigital web of the foot, the 'toe pinch' reflex, produces a variable response. In one study, surgical anaesthesia was ascertained by closed eyelids, mydriatic pupils, delayed pupillary light reflex, slow movement of the nictitating membrane across the cornea, relaxed muscles and absent pain reflexes (Korbel, 1993).

BOX 9.2 Reflex responses at a surgical plane of anaesthesia (Korbel, 1993)

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Please refer to the printed publication.

Temperature

Thermoregulation is reduced in anaesthetised patients and core body temperature should be monitored during anaesthesia. Animals suffering from hypothermia will additionally be more susceptible to cardiac instability, require less anaesthetic and have a more prolonged recovery (Edling, 2005). It is, thus, vital to minimise heat loss during anaesthesia and provide supplemental heating to patients.

Small probes attached to a digital thermometer can be used to record cloacal temperature (Coles, 1997). This will enable monitoring of the bird's core body temperature during anaesthesia. The probe is carefully placed in the cloaca, but readings may vary with body position and cloacal activity. Oesophageal thermistor probes are more accurate, with the flexible probe inserted to the level of the heart (Edling, 2006). If the patient is too small or a suitable probe is unavailable, placing a thermometer between the patient and heat pad will provide an estimate of the bird's temperature.

A drop in body temperature of more than 5.5°C will cause circulatory problems and a prolonged recovery time (Forbes and Altman, 1998). Normal body temperature should return rapidly after the end of anaesthesia, although supplemental heat is usually required.

Anaesthetic monitoring equipment

An 8 MHz *Doppler probe* can be used to assess pulse rate and rhythm in the brachial or dorsal metatarsal arteries. As the volume output from the Doppler is related to the strength of the pulse, this will effectively assess peripheral circulation and thence cardiac function.

ECGs can be used, but many machines designed for larger animals will not record the high rates (up to 500 bpm) seen in avian patients. The Silogic EC-60® (Silogic International Ltd, Surrey, UK) can be used in birds and will

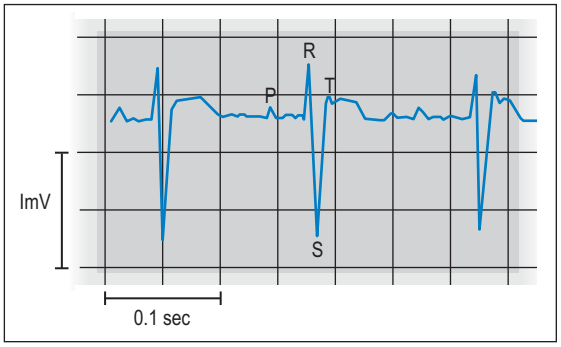


Figure 9.13 • Schematic avian ECG (lead II, 100 mm/s, 10 mm/mV). (After Schoemaker and Zandvliet, 2005)

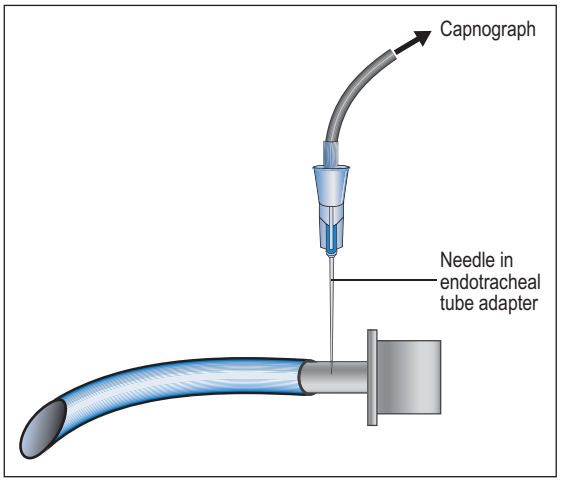


Figure 9.14 • Side stream sampling for capnography with minimal dead space.

also record respiratory rate (Coles, 1997). ECGs are useful to assess for arrhythmias, conduction disorders and metabolic disorders (Rosenthal and Miller, 1997).

It can be difficult to connect ECG leads to avian skin, although the use of alcohol or gel is helpful. ECG pads can be attached to relatively featherless areas of skin, such as the ventral propatagium or inguinal skin (see Fig. 9.8). The leads are connected as for other species, using the wings and legs (Lumeij and Ritchie, 1994). Alternatively, an oesophageal ECG probe (Cardio Companion ECG Probe and Esophageal Lead, SurgiVet Inc, Waukesha, WI) can be used in avian patients with an adaptor connecting the leads to the oesophageal probe (Edling, 2006).

The ECG produced is different to other species and between avian species (Fig. 9.13) (Casares et al., 2000; Lopez Murcia et al., 1995; Lumeij and Ritchie, 1994; Uzun et al., 2004). A sinus rhythm is normal in birds. Gender differences in the ECG may be found (Lopez Murcia et al., 1995; Rodriguez et al., 2004). Reference ranges exist for a few species, but will vary depending on positioning, whether the patient is conscious or anaesthetised, and between anaesthetic protocols (Casares et al., 1999; Lumeij and Stokhof, 1985; Nap et al., 1992; Schoemaker and Zandvliet, 2005).

If the ECG is used purely for anaesthesia monitoring, two leads may be used to the sternum across the cardiac axis; one lead is placed cranially and paramedially on the right, with the other caudal and slightly to the left (Pees et al., 2006).

Direct arterial *blood pressure* measurement is difficult in birds due to their small size. However, indirect measurements can be obtained by using inflatable cuffs, a Doppler blood pressure monitor and a sphygomomanometer (Lichtenberger, 2005).

A *pulse oximeter* can be used to monitor arterial oxygen saturation, but readings are not consistently accurate in birds (Schmidt et al., 1998). Oximeters can be used to monitor trends during anaesthesia. The level should ideally be more than 90%. Levels below 80% are dangerous for the patient (Coles, 1997).

Arterial partial pressure of oxygen (PaO_2) and oxygen saturation of haemoglobin do not appear to be sensitive indicators of the ventilatory status in birds, at least not in those receiving 100% inspired oxygen, which most patients do during anaesthesia (Edling et al., 2001; Schmidt et al., 1998). Oxygen saturation may be normal even in hypercapnic animals (Eger, 1993). The partial pressure of arterial oxygen, PaO_2, may be significantly affected by problems such as apnoea, ventilation–perfusion mismatch and tracheal obstruction (Edling, 2005).

Blood gas analysis, including assessment of arterial oxygen and carbon dioxide, is the gold standard for assessment of respiratory function during anaesthesia. However, this is usually impractical owing to technical problems with arterial blood sampling, small patient size prohibiting repeat sampling or financial considerations.

Capnography is commonly used to assess the end-tidal carbon dioxide ($P_{ET}CO_2$) of the anaesthetised patient. Capnograph machines are becoming more widely available in veterinary practices and their use is discussed in the introductory chapter. Capnographs have been trialled in various avian species (Desmarchelier et al., 2007; Edling et al., 2001). It is important to avoid introducing extra dead space to the anaesthetic circuit in small animals and to avoid extra resistance within the circuit. Capnographs that sample from the side of the circuit are, therefore, preferable to those which sample within the stream of gases. The capnograph can be attached to the endotracheal tube adapter via an 18-gauge needle, with the bevel towards the patient (Fig. 9.14). Avoid obstructing the airway with the needle (Edling, 2006).

Capnography can be used to assess ventilation in birds as an indirect measure of arterial carbon dioxide. $P_{ET}CO_2$ correlates reasonably well with $PaCO_2$. However, the anatomy and physiology of the avian lungs create an efficient cross-current exchange system, which produces a higher concentration of carbon dioxide in expired gases (end-tidal partial pressure of carbon dioxide, $P_{ET}CO_2$) than in the arteries (arterial partial pressure of carbon dioxide, $PaCO_2$) (Brackenbury, 1987; Davies and Dutton, 1975). A study in African grey parrots (*Psittacus erithacus*) showed

that $P_{ET}CO_2$ overestimated $PaCO_2$ by approximately 5 mmHg (Edling et al., 2001).

In order to maintain a normal acid–base balance in anaesthetised birds, the recommendation is to perform PPV to produce $P_{ET}CO_2$ measurements between 35 and 45 mm Hg (Desmarchelier et al., 2007; Edling, 2001).

Respiratory monitors can be used, but care should be taken not to introduce extra resistance or dead space to the anaesthetic circuit. For example, the Imp respiratory monitor (IMP Electronics, Oxford Medical Systems, UK) or an apALERT apnoea monitor (MBM Enterprises, Australia) may be used in birds (Coles, 1997).

PERI-ANAESTHETIC SUPPORTIVE CARE

Fasting

The risks associated with not fasting a bird relate to regurgitation during anaesthesia, with the possibility of acute respiratory obstruction and death or of inhalation pneumonia. Due to the high avian metabolism, the almost continuous requirement for energy supply, poor hepatic glycogen storage and the probability of malnutrition on presentation, birds should not be fasted for long periods before anaesthesia. The crop should be palpated before anaesthesia in all patients to check for food or liquid residues. If material is present within the crop, it can be aspirated before induction of anaesthesia (Edling, 2006). After induction, the head should be raised above the level of the crop to prevent regurgitation.

Birds of prey eat once or twice daily, and should be fasted for 12 h to allow the previous meal to be digested and a cast produced before induction of anaesthesia. Alter-natively, food can be fed without casting material (for example, meat without fur/feathers and bones) on the previous day. Most granivorous or omnivorous species eat throughout the day and are more likely to become hypoglycaemic if fasted. Birds between 300 g and 1 kg can be fasted for up to 6 h and those between 100 g and 300 g for 3 or 4 h. Birds weighing less than 100 g should never be fasted (Coles, 1997).

- Do not fast neonates or birds that weigh less than 100 g before anaesthesia.
- The total period without food includes the planned pre-anaesthetic fasting and the post-anaesthetic period before the patient is eating again. Assist feeding may be required in birds that do not begin eating soon after anaesthesia.

Oxygen

Birds with respiratory compromise or anaemia will benefit from supplemental oxygen before and after anaesthesia. Anaesthesia will depress the respiratory system and all anaesthetised patients should be provided with oxygen via facemask, endotracheal tube or air sac cannula.

Heating

As anaesthetics reduce the patient's thermal homeostatic mechanisms, supplemental heating should be provided. This may be in the form of insulation to prevent heat loss, such as towels, blankets or bubble-wrap. It is usually necessary also to have a heat source by the patient, for example warm air blanket (for example, the Bair Hugger®, Arizant Inc, Eden Prairie, MN), hot water bottle, circulating water blanket, heated surgery table, electric heat pad or heat lamp. Forced warm air blankets are particularly effective at minimising heat loss (Edling, 2006). The patient's eyes should be protected from drying by warm air devices with lubrication, for example liquid paraffin. It is important to avoid overheating and contact burns, and a towel should be placed between the heat source and patient, particularly over featherless areas. Radiant heat sources appear to be the most effective way of providing heat (Edling, 2005). Background heating of the room is useful. It is useful to pre-warm an incubator (see Figs 9.5 and 9.6) ready for recovery post-anaesthesia.

Evaporation from mucous membranes is a significant source of heat loss in anaesthetised patients. If possible, anaesthetic gases should be warmed and humidified (Korbel, 1993). Fluids should also be warmed before administration.

As discussed above, the patient's body temperature should be monitored closely during the procedure. Hypothermia is more common, but hyperthermia may also occur if excessive attempts at supplementing heat are employed.

Fluids

Fluids should be administered, preferably intravenously, during anaesthetics longer than 20 min. In most patients, a bolus of fluids of 10–20 ml/kg is sufficient to replace fluid lost during anaesthesia and to speed recovery (Forbes and Altman, 1998). During more prolonged anaesthetics or those in critical patients, intravenous access with a continuous rate infusion is more appropriate.

It is particularly important to maintain blood pressure if anaesthetic agents are renally excreted. Routinely, Hartmann's solution or lactated Ringer's is given. If hypoglycaemia is considered a risk, glucose may be added to the crystalloids; this also assists hepatic detoxification of drugs (Coles, 1997). The optimum route for fluid administration is intravenous or intraosseous. Although absorption is slower, subcutaneous fluids are easier to administer. Fluids should be warmed to body temperature prior to administration, to reduce the risk of hypothermia.

Analgesia

Analgesia is important for many procedures, and the lack of provision of appropriate analgesia is likely to affect not only the avian patient's immediate condition, but also its recovery. It may be difficult to ascertain whether a bird is affected by pain, as signs may be subtle. Behaviours associated with

pain in birds depend on the individual bird, and the location and type of pain. The bird may show specific behaviours, such as lameness, guarding a painful area or vocalisation; alternatively it may show more general abnormalities in behaviour, for example inactivity, lack of grooming or reduced appetite (Paul-Murphy, 2006). If a bird has a condition that causes pain in other species or will undergo surgery, the veterinary surgeon has an ethical duty to provide analgesia. Intra-operative analgesia may also reduce the doses of anaesthetic agents required, reducing the side effects and risks associated with anaesthesia.

As with other species, pre-emptive analgesia appears to reduce pain (Woolf, 1994; Woolf and Chong, 1993). Minimising environmental stressors can also reduce pain in avian patients (Paul-Murphy, 2006). Certain anaesthetic agents will have analgesic properties, such as alpha-adrenergic agonists and ketamine. If alpha-adrenergic agents are reversed, their analgesic properties are also lost. If anaesthetics without analgesic effects are used, the analgesic agents listed below should be employed. In fact, multimodal analgesia is preferable; using lower doses of more than one agent to reduce possible side effects associated with individual agents.

Local anaesthetics

Local anaesthetics may be used in birds to provide analgesia, although rarely as the sole agent for a procedure as conscious restraint and handling of the patient are likely to cause stress (Edling, 2006). These drugs block the transmission of noxious impulses, producing regional anaesthesia and analgesia (Paul-Murphy, 2006). As with other analgesics, administration of local anaesthetics pre-emptively produces much more effective analgesia.

These agents have long been associated with toxicities in avian species, as overdosage is easy in small patients and systemic uptake appears to be rapid. Signs of toxicity include tremors, ataxia, recumbency, seizures, stupor and cardiovascular effects, including cardiac arrest and death (Paul-Murphy, 2006). However, local anaesthetics can be safely used if the dose is calculated based on an accurate body weight. It may be useful to dilute the solution. It is also helpful to calculate the maximum safe dose and only draw up this volume before administration. The duration of action of local anaesthetics is not known in most avian species.

- Overdosage of local anaesthetic is easy and can be fatal.
- Calculate the dose based on an accurate body weight.
- The toxic dose of lidocaine (lignocaine) in birds is approximately 4mg/kg.

Local anaesthetics are commonly administered topically. Benzocaine has been used for minor wound repair (Clubb, 1998). Bupivacaine provided 4 h of analgesia in chickens after beak trimming (Glatz et al., 1992). Creams and ointments containing local anaesthetics pose two problems, namely those of dose calculation and of plumage damage, and are not recommended in avian species.

Lidocaine (lignocaine) can be used in birds, although for smaller patients it is safer to use a less concentrated solution, for example by diluting a 2% solution to 0.2% solution. The addition of adrenaline (epinephrine) will reduce the rate of absorption, but some texts recommend use of preparations that do not contain adrenaline (epinephrine) (Paul-Murphy, 2006). The toxic dose of lidocaine (lignocaine) in birds is around 4 mg/kg (Huckabee, 2000; Paul-Murphy and Ludders, 2001).

Bupivacaine has been safely used at 1 mg/kg in birds (Paul-Murphy, 2006). Local anaesthetics may also be injected into joints to provide local analgesia. One study reports the use of intra-articular bupivacaine in birds with experimentally induced arthritis (Hocking et al., 1997). Procaine has a narrow safety margin, so should be avoided in small birds and diluted for larger birds (Coles, 1997).

Opioids

Opioid agents may be used as part of the anaesthetic protocol to reduce the doses of other agents required, and most additionally provide good analgesia. The distribution and function of opioid receptors vary between species, producing different susceptibilities to different opioids. For example, pigeons have been shown to have high proportions (76%) of kappa (κ) opioid receptors in their forebrain (Mansour et al., 1988). Species differences mean that different agents will produce differing effects on receptors. For example, the κ agonist butorphanol had an isoflurane-sparing effect in cockatoos (*Cacatua* spp.) and African grey parrots (*Psittacus erithacus*), but not in Amazon parrots (*Amazona* spp.) (Curro, 1993; Curro et al., 1994).

Dose rates necessary to provide analgesia may also vary between avian species (Paul-Murphy et al., 1999). Other issues to address are that plasma levels necessary for analgesia in other species may not coincide with those required in avians and that plasma half-life of drugs may be much shorter in birds. The high doses required to produce analgesia in these animals may produce severe side effects, for example fentanyl at analgesic doses produced hyperactivity in cockatoos (Paul-Murphy and Ludders, 2001). Side effects commonly seen with opioid use include cardio-respiratory depression, for example reduction in heart rate and tidal volume (Paul-Murphy, 2006). Butorphanol administered before anaesthesia with sevoflurane did not cause significant changes in anaesthetic and cardiopulmonary parameters in a study in Hispanioloan Amazon parrots (*Amazona ventralis*) (Klaphake et al., 2006).

Non-steroidal anti-inflammatory drugs

NSAIDs can be utitilised as anti-inflammatories and analgesics in avians. The roles of prostaglandins in inflammation and pain physiology are similar in birds to those in other species (Nichol et al., 1992). However, the relative expression of cyclooxygenase-1 (COX-1) and COX-2 enzymes requires further elucidation. Pre-emptive administration of NSAIDs reduces peripheral sensitisation to noxious stimuli (Edling, 2005).

Table 9.7: Analgesics for avian species

DRUG	SPECIES	DOSE (mg/kg)	ROUTE	DOSING INTERVAL (hours)	COMMENT
Local anaesthetics:					
Bupivacaine	Studies in ducks and chickens	2[12,15]	SC, intra-articular, topical IM, SC	Not known (4–6 h in mammals)[6]	Dilute 1:10 or more before administration[16]
Lidocaine (lignocaine)	All	1.0–3.0 [1,10,18]		(90–200 min in mammals)[6]	
Steroids:					Anti-inflammatory, shock, trauma
Dexamethasone	Most species	0.2–4.0 [2,10,20]	IM, IV	12–24	May affect immune competency
Non-steroidal anti-inflammatory drugs:					
Carprofen	All	2.0–4.0 [5,7,10,14,18]	PO, IM, SC, IV	8–12	Doses <10 mg/kg reported
Flunixin	All	0.5 [9]	IM	24	Ensure hydrated – potential nephrotoxicity
Ketoprofen	All	2.0 [4,8,12,18]	PO, SC, IM	8–24[7]	
Meloxicam	Psittacines, raptors	0.1–0.2 [4,18,19]	PO, IM	12–24	
Piroxicam	Most	0.5 [7,18]	PO, IM PO	12	Chronic pain, e.g. osteoarthritis
Opioids:					
Buprenorphine	Most species	0.01–0.05[11]	IM	8	Mixed agonist–antagonist
Butorphanol	Most species	0.5–0.75 [1,17]	IM, IV	12	Mixed agonist–antagonist, primary kappa agonist action
Fentanyl	Cockatoos	0.2[18]	SC	–	May have excitement phase
Morphine	–	–	–	–	Appears ineffective as analgesic in many avian species

Key: IM = intramuscular, IV = intravenous, PO = oral, SC = subcutaneous

[1] (Abou-Madi, 2001); [2] (Bauck, 1993); [3] (Carpenter, 2005); [4] (Coles, 2001); [5] (Cooper, 2002); [6] (Cornick, 2001); [7] (Edling, 2005); [8] (Graham et al., 2001); [9] (Heard, 1997); [10] (Huckabee, 2000); [11] (Jenkins, 1993); [12] (Machin and Livingston, 2001); [13] (Malley, 1994); [14] (Moore and Rice, 1998); [15] (Mulcahy and Tuomi, 2001); [16] (Paul-Murphy, 2006); [17] (Paul-Murphy et al., 1999); [18] (Paul-Murphy and Ludders, 2001); [19] (Stanford, 2002); [20] (Tully, 2000)

A few studies have shown NSAIDs to be effective in treatment of pain in several avian species (Danbury et al., 2000; Machin et al., 2001; McGeown et al., 1999). For example, ketoprofen at 5 mg/kg was shown to reduce the response to noxious stimuli in Mallard ducks (*Anas platyrhynchos*) anaesthetised with isoflurane (Machin and Livingston, 2002). However, little is known about therapeutic plasma levels or toxic doses and most published doses are empirical.

EMERGENCY PROCEDURES/DRUGS

The section above discusses some problems that may arise during anaesthesia in avian species and how to avoid them. Careful patient preparation and monitoring during anaesthesia will greatly reduce the incidence of problems with avian anaesthesia. This section will focus on crises that may arise and how to manage them.

Many of the procedures for which avian species require anaesthesia are non-invasive, for example phlebotomy and radiography. If the patient is not responding well under anaesthesia in these situations, the clinician should be prepared to postpone the procedures and recover the patient. Unless a flock problem is suspected and the owner is prepared for the individual bird to be sacrificed there is no point in making a clinical diagnosis if the patient dies during the investigations.

Preparation before induction

As with other species, preparation of drugs and equipment before induction will minimise the period for which the animal remains under anaesthesia. Preparation is vital as, the longer the patient is anaesthetised, the more likely problems will occur. A 'crash kit' (see Fig. 1.8) should always be on hand during anaesthesia, stocked with appropriate endotracheal tubes, intravenous catheters and emergency drugs. It is wise to check before each anaesthetic that the kit contains equipment likely to be used, for example an appropriately sized endotracheal tube for the patient. This will minimise the time before emergency treatment can be initiated, improving the patient's chances of recovery.

Time is of the essence when dealing with avian anaesthetic emergencies, as respiratory arrest is often rapidly followed by cardiac arrest. The patient will have a greater chance of survival if emergency equipment and drugs are close at hand should the need arise. If a patient is unwell prior to anaesthesia, or a major procedure is to be performed, it is advisable to calculate emergency drug doses beforehand. Doses for emergency drugs can even be drawn into syringes before anaesthesia, in readiness for potential problems (and discarded after the procedure if not required). During long procedures, it is advisable to have access to the cardiovascular system, and an intravenous or intraosseous catheter should be placed after induction. This will allow provision of fluids during the procedure and immediate access to the circulation if required.

Problems encountered in avian anaesthesia

The most commonly encountered problems in anaesthesia of avian patients are apnoea, hypoventilation, hypothermia and regurgitation (Edling, 2006). Prevention of problems is obviously better than treatment. Careful assessment and stabilisation of the patient, selection of anaesthetic protocol and attentive monitoring of the patient will reduce the likelihood of these problems occurring.

Regurgitation

This may occur at any point during the anaesthetic. However, considerations before and at the onset of anaesthesia should reduce the risk of this happening. As discussed above, birds larger than 100 g are usually fasted for a short period prior to anaesthesia to allow the crop to empty. The crop should be palpated before induction to assess for the presence of ingesta. If fluid is present, it is advisable to aspirate crop contents as fluid is likely to be regurgitated during anaesthesia. Endotracheal intubation after induction with an appropriately sized tube will help protect the airway should regurgitation occur. The patient should be positioned so that the neck is elevated, to reduce the risk of regurgitation during anaesthesia, and the beak is pointing down, to allow drainage should any regurgitation occur.

If regurgitation occurs, suction and swabs can be used to clear the oral cavity and airway. If the patient is not intubated, an endotracheal tube should be placed. If aspiration is suspected, broad-spectrum antibiotics should be initiated as pneumonia is likely to ensue. The patient should be closely monitored for signs of respiratory disease in the post-operative period, further medication administered as appropriate if signs develop and a guarded prognosis given.

Respiratory problems

As discussed above, *positioning* during anaesthesia is important. Dorsal recumbency should be avoided if possible due to the compression of the caudal thoracic and abdominal air sacs by the abdominal viscera, reducing their effective volume. Although gas exchange within the lungs will not be affected, the bellow movements of air caused by the air sacs will be severely limited. Ventilation of birds in ventral recumbency is also abnormal, as sternal movements are restricted.

IPPV should thus be performed on patients in dorsal and ventral recumbency to maintain adequate ventilation (Edling, 2005, 2006). Lateral recumbency is preferable to sternal recumbency. The wings should not be stretched excessively, as brachial plexus nerve damage may result and tight restraint may limit respiratory movements (Coles, 1997).

Most anaesthetics will produce some *respiratory depression*, but if the respiratory rate or depth is significantly reduced, action should be taken. This may be due to an overdose of anaesthetic. Respiratory depression may also cause hypercapnia; capnography can be used to detect this. If prolonged respiratory depression occurs, the concentration of volatile agent should be reduced or switched off, the oxygen flow rate increased and PPV performed to wash out anaesthetic and carbon dioxide from the air sacs.

Unless PPV is used to counteract respiratory depression during anaesthesia, the $PaCO_2$ will rise, potentially fatally (Marley and Payne, 1964). High gas flow rates will help remove waste gases, but this may not be sufficient if the patient is unable to eliminate carbon dioxide efficiently, for example with a reduced respiratory rate. The *hypercapnia* will produce respiratory acidosis, myocardial depression and hypotension. Elevated $PaCO_2$ also predisposes the heart to atrial and ventricular fibrillation, and cardiac failure (Coles, 1997).

During anaesthetic induction, periods of apnoea and bradycardia may occur as the physiological '*dive response*'.

This is particularly common with mask induction in waterfowl, and the reader is referred to other texts for anaesthetic peculiarities with these species (Beynon et al., 1996). If the patient becomes apnoeic during mask induction, the mask should be removed and oxygen provided via an open mask (avoiding stimulation of the beak and nares which elicits the response) until respiration resumes (Edling, 2006).

There are several reasons why *respiratory arrest* may occur during avian anaesthesia, including hypercapnia, airway obstruction, primary cardiac arrest and the end-point of other respiratory problems. However, the most common cause is an anaesthetic overdose (Coles, 1997).

Close patient monitoring should identify early signs of patient compromise and allow correction of problems before apnoea occurs. However, if the patient becomes apnoeic, rapid assessment of the anaesthetic equipment and response is imperative. As with respiratory depression, the anaesthetic agent should be switched off in the case of volatile agents or reversed in the case of injectables. If the patient is not already intubated this should be performed, if possible, and PPV commenced. If intubation is not possible, for example if the patient is too small, PPV can still be performed via a close-fitting facemask.

During PPV care should be taken not to overinflate as this may cause damage to the delicate airways. Overventilation may also wash out carbon dioxide, inhibiting the chemoreceptors needed to stimulate respiration (Coles, 1997). If an oxygen supply is not available, it is still prudent to intubate and provide PPV using room air, for example using an Ambu-bag or resuscitator (see Fig. 1.12). If mechanical ventilation is not possible, movement of the wings and keel may stimulate respiration.

Equipment should be checked for abnormalities, such as an obvious obstruction in the circuit. The patient should be assessed for other responses, such as heart rate, reflexes and spontaneous respiratory movements. If spontaneous respiration does not commence within 2 or 3 min, doxapram can be given to stimulate respiration. This acts directly on respiratory centres in the medulla, stimulating ventilation (Edling, 2006). This can be administered intravenously, intramuscularly, intratracheally or directly on to mucous membranes in the oral cavity. As the duration of action is short, the dose may need to be repeated.

Apnoea, ventilation–perfusion mismatch and tracheal obstructions will affect the partial pressure of arterial oxygen, even when 100% oxygen is administered to patients. As arterial blood gas analysis is not usually an option and pulse oximetry is unreliable in birds, $P_{ET}CO_2$ should be closely monitored as an indication of ventilation (Schmidt et al., 1998).

Periods of apnoea can rapidly cause serious metabolic derangements in avian patients. Their small FRC means that gas exchange will not occur without sufficient airflow through the lungs. Normal maintenance of the acid–base balance by the respiratory system also ceases during apnoea (Edling, 2006). IPPV throughout anaesthesia will reduce this problem by ensuring adequate ventilation of the respiratory tract during the procedure.

Cardiac arrest often follows respiratory arrest. Therefore, a rapid response is vital if apnoea is detected and cardiovascular parameters should be closely monitored during apnoea. With most avian anaesthetics, there is a time lag after respiratory arrest before cardiac arrest. However, this may not be seen with halothane anaesthesia.

Airway obstruction is common when birds are anaesthetised for prolonged periods, as anaesthetic gases dehydrate airway secretions and respiratory depression means that secretions are not mobilised; airway *secretions* may then obstruct the respiratory tract or endotracheal tube. Small birds are especially at risk due to the narrow diameter of their airways and endotracheal tubes used. As airway occlusion occurs, the expiratory phase of respiration is prolonged (Ludders and Mathews, 1996). Secretions are less likely to obstruct the airway if PPV is performed, as this assists in moving secretions so they do not build up (Edling, 2006).

Signs seen may include an increase in respiratory movements or noises, such as clicking, gurgling or squeaking. Cyanosis is rare in birds, but if seen carries a poor prognosis (Coles, 1997).

If the bird is intubated, remove the endotracheal tube and replace it with an unobstructed one (or clean the blocked tube before replacement). Placement of an air sac cannula is another option for administration of gases to the respiratory tract. This is particularly useful where intubation is not possible or if lower airway obstruction is present. Check the oropharynx for any other secretions that may be inhaled, using suction or cotton buds to remove them.

Atropine is not routinely used in avian anaesthesia. Its usefulness for reducing respiratory secretions should be balanced against the increase in viscosity of salivary, tracheal and bronchial secretions that are produced, which will increase the risk of airway obstruction (Edling, 2006).



Figure 9.15 • Tape wrapped around the endotracheal tube and beak will hold the tube in place, as in this Harris' hawk (*Parabuteo unicinctus*).

Atropine may also inhibit $PaCO_2$ chemoreceptors, and may lead to respiratory depression (Coles, 1997).

Cardiovascular problems

Most anaesthetics reduce *blood pressure* (Coles, 1997). If a drop in blood pressure is detected, a bolus of fluids should be administered. Initially crystalloids may be used, but if there is no response or the hypotension is severe, colloids may be more appropriate. The blood pressure should, therefore, be monitored during anaesthesia, usually via an indirect method.

Hypercapnia is the most likely cause of *cardiac failure* during avian anaesthesia. Cardiac failure is often multifactorial, and other factors include hypoxia, anaesthetic agent, dehydration, hypothermia and poor positioning (Coles, 1997).

In most cases, asystole is preceded by bradycardia. If bradycardia is noted, the concentration of volatile anaesthetic agent should be lowered and the parasympathetic agent atropine may be administered to correct this (Edling, 2006). Cardiac arrest may rapidly follow respiratory arrest during avian anaesthetics, particularly if an agent such as halothane is used when respiratory and cardiac arrest occur simultaneously.

If asystole occurs, anaesthetic agents should be turned off and reversal agents administered for injectable anaesthetics. Airway patency should be checked and 100% oxygen administered via IPPV. Adrenaline (epinephrine) can be administered intravenously or intratracheally. Intravenous lidocaine (lignocaine) may also be useful. Drugs administered intravenously should be followed by a bolus of fluids to assist their passage to the heart and also to increase blood pressure. If an intravenous catheter has not been previously placed, venous access is likely to be extremely difficult if not impossible in the collapsed circulation. Intracardiac injection can be attempted, but is difficult due to the protection of the heart within the rib cage and sternum. External cardiac massage can be attempted, but is similarly difficult. Internal cardiac massage can be performed using a sterile gloved finger or sterile cotton bud via a coelomic incision, but is rarely successful (Coles, 1997).

- Always have an emergency or 'crash' kit on hand when anaesthetising birds.
- Ensure it is kept well stocked.
- Calculate drug doses for patients beforehand.

Hypothermia

As with other animals, anaesthetised birds lose their thermoregulatory abilities, particularly small patients with a large surface area to volume ratio. Dry anaesthetic gases and surgical skin preparations increase heat loss. Supplemental heating should, therefore, be used to avoid hypothermia in anaesthetised birds. Hypothermia will

Table 9.8: Approach to avian anaesthetic emergencies

CHECK	TECHNIQUE
Airway	Intubate or cannulate air sac
Breathing	Provide intermittent positive pressure ventilation (manual or mechanical ventilator) – preferably via endotracheal tube or air sac cannula, but possible via close-fitting facemask
Circulation	Obtain intravenous or intraosseous access 10 ml/kg bolus of Hartmann's External cardiac compression not usually effective; internal cardiac massage possible (often unsuccessful)
Drugs	See Table 9.9

(Adapted from Edling, 2005)

Table 9.9: Emergency drugs in birds

DRUG	DOSE	ROUTE	COMMENT
Adrenaline (epinephrine) (1:1000)	0.5–1.0 ml/kg [1,4]	IV (IC), IM, IO, IT	Treatment of cardiac arrest. (Can dilute with Hartmann's to calculate dose)
Atropine	0.2–0.5 mg/kg [2,4]	IV, IM, IO, IP	Treatment of supraventricular bradycardia; CPR
Diazepam	0.5–1.0 mg/kg [3]	IM, IV	Seizures. Repeat until response seen
Doxapram	5–20 mg/kg [1, 5]	IV, IM, IO, IT, Top	Respiratory depression or arrest

Key: CPR, cardiopulmonary resuscitation, IC = intracardiac, IM = intramuscular, IO = intraosseous, IT = intratracheal, IV = intravenous, Top = topically

[1] (Carpenter, 2005); [2] (Redig, 1998); [3] (Rupiper and Ehrenberg, 1994); [4] (Rupley, 1997); [5] (Huckabee, 2000)

reduce metabolism of anaesthetic agents and other drugs administered to the patient, and will depress the myocardium, resulting in cardiac instability (Edling, 2006). Hypothermic animals require more energy reserves to generate heat during recovery. At the very least hypothermia will lead to a prolonged recovery; a drop in core temperature of 5°C may be fatal (Coles, 1997).

Dehydration

This may result from prolonged anaesthesia, as this may allow fluids to be lost from the air sacs. This is particularly serious in patients that are already dehydrated. The reduction in circulating blood volume will produce a decrease in cardiac output, with ensuing reduced tissue perfusion. Anaerobic respiration may result in metabolic acidosis (Coles, 1997).

Ocular damage

The eyes may become dry or be traumatised unless they are protected during anaesthesia. Lubricants are applied after induction and care taken not to place the eye directly on to a hard or abrasive surface.

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10 Passerine, psittacine and columbiforme anaesthesia

INTRODUCTION

This chapter discusses anaesthesia in pet birds belonging to three orders – Passeriformes, Psittaciformes and Columbiformes. Although there are many differences between species, the approach to their anaesthesia is similar. The preceding chapter covers most aspects of this approach. The aim of this chapter is to highlight anatomical, physiological and pathological differences within these three groups to allow the clinician to refine anaesthetic techniques. Where studies have been performed on one species, this will be highlighted, as will any other differences affecting anaesthesia.

The order Passeriformes includes over 5000 species. Passerines commonly seen in veterinary practice include species such as canaries and zebra finches. Pet Psittaciformes include the Cacatuidae (cockatoo) and Psittacidae (parrot) families. Psittacidae include budgerigars and cockatiels as well as the larger parrots. Pigeons and doves belong to the Columbidae family in the Columbiformes order.

ANATOMY AND PHYSIOLOGY

Metabolism

Passerines have the highest metabolic rate of all vertebrates (Dorrestein, 1997; Maina, 1996).

Cardiovascular system

The left jugular vein is usually much smaller than the right (Akester, 1971; West et al., 1981), and may be absent in some small cage bird species (Evans, 1996; King and McLelland, 1984). Pigeons have an extensive vascular plexus in the neck that makes the jugular an unsuitable vein for phlebotomy in these species (Coles, 1997).

Several aspects of pet psittacine husbandry may affect the bird's response to anaesthesia. Atherosclerosis results from cholesterol deposition in arteries and is associated with inflammation and fibrosis, and it is common in captive psittacines (Johnson et al., 1992). Many pet parrots are fed unbalanced diets, high in fats that predispose to obesity and these atheromatous arteries (Coles, 1997). This is exacerbated by a sedentary lifestyle, with many captive birds having little opportunity for free flight to maintain fitness.

Respiratory system

In psittaciformes, the right and left nasal sinuses communicate (Evans, 1996; King and McLelland, 1984). Access for intubation is more difficult in psittacines due to their fleshy tongue obscuring the glottis (Fig. 10.1) (O'Malley, 2005).

Most (88–90%) of the lungs in pigeons are paleopulmonic and ordered in parallel; thus the majority of air flow through them will be unidirectional (Edling, 2006; Ludders and Mathews, 1996).

Passeriformes have a total of seven air sacs, as the cranial thoracics are fused with the clavicular (Evans, 1996; King and McLelland, 1984). Pneumonisation of the medullary cavity of bones is minimal in some small birds. Due to the large internal surface area of the air sacs and the high body temperature of small birds, they lose a substantial amount of water via the respiratory tract (Coles, 1997).

Urinary system

The nasal glands are absent or vestigial in passerines (Shoemaker, 1972). Species such as the zebra finch, *Taeniopygia guttata*, can produce metabolic water and concentrated urine (Goldstein and Skadhauge, 2000).

Digestive system

Gastrointestinal transit times vary from 16 min to 2 h in passerines (O'Malley, 2005). They also have high metabolic

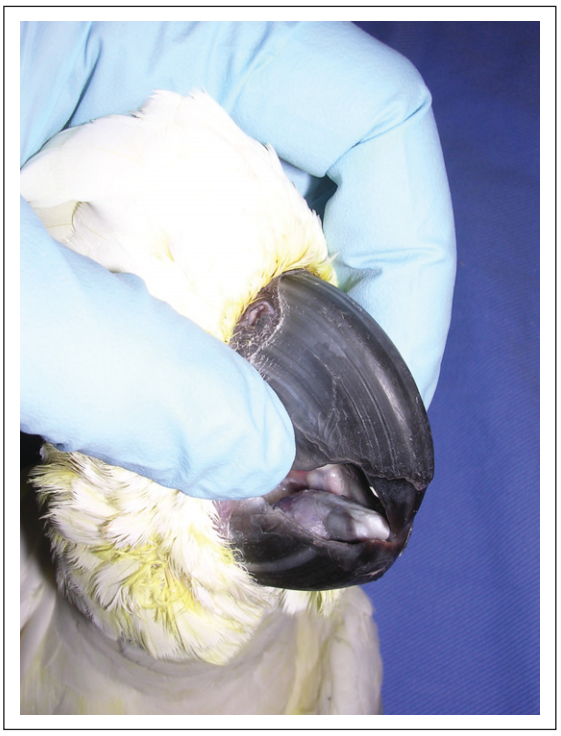


Figure 10.1 • Oral cavity of a sulphur-crested cockatoo (*Cacatua galerita*) showing the typical fleshy tongue seen in Psittaciformes.

demands, and hypoglycaemia is common in birds that have been malnourished or anorexic even for a short period of time (Harrison et al., 2006). Fasting is not usually recommended before anaesthesia in these small birds. The rapid gastrointestinal transit time means the crop will empty quickly.

Diets vary between species. Passerines feed mainly on seeds and/or insects in the wild, and most pet birds will take small quantities of fruit and vegetables. Formulated diets are available for many species (for example, Harrison's High Potency Formulas®, *http://www.harrisonsbirdfoods.com/*, or Kaytee® exact®, Kaytee Products Inc., Chilton, WI).

As discussed in the general section, all-seed diets will cause immune depression, and associated respiratory and/or renal disease is possible (McDonald, 2006). Obesity may also lead to cardiac disease and hepatic lipidosis (McDonald, 2006). All these factors increase the risk associated with anaesthetising these birds. Most psittacines presented to veterinary surgeons are house- or cage-bound pets. In general, they will be unfit, particularly those that have their wings clipped or do not fly for some other reason. In combination with this, many are on an unbalanced diet, with excess fats and vitamin deficiencies.

Although a bird's current diet may be unsuitable, it is unwise to attempt to change the diet during hospitalisation of a sick bird. The bird is more likely to eat if it is offered food that it recognises.

Endocrine system

Pituitary gland neoplasia is common in budgerigars (*Melopsittacus undulates*). Resulting changes to prolactin and vasotocin production may cause hyperglycaemia and polydipsia or polyuria; pressure from the lesion may cause blindness, exophthalmos and seizures (Rae, 2000).

Goitre due to iodine deficiency is also common in budgerigars (Oglesbee et al., 1997). Thyroid disease may affect metabolism, growth, reproduction and thermoregulation (Hodges, 1981; King and McLelland, 1984; Rae, 2000).

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

It is vital to obtain husbandry details for pet birds, as poor husbandry will predispose birds to debility and other conditions (discussed below) that will increase anaesthetic risks. Many pets, particularly psittacines, are prevented from flying either by wing clips or small enclosures, and are in a state of cardio-respiratory unfitness when presented to the clinician. Pigeons are often kept in captivity for racing and are more likely to be cardiovascularly fit.

Very small birds are gently restrained for examination with the clinician's hand around their body, avoiding compression of the keel that would preclude respiratory movements, with the head between two fingers or thumb and forefinger. A tissue or thin cloth may be lightly wrapped around the patient. Restraint of psittacines is similar to passserines, but a towel is usually necessary to wrap the wings in these larger species (and has the added benefit of providing the bird with something to chew other than the clinician's fingers). With larger species, such as macaws, an experienced assistant should restrain the patient to enable the veterinary surgeon to conduct the clinical examination. Most captive pigeons are accustomed to handling and, therefore, are less stressed during restraint than the other species.

The examination should concentrate on identification of factors that may affect anaesthesia, such as increased or altered noise on air sac auscultation. Due to the delicate avian respiratory system and susceptibility to infectious or toxic agents, all birds presented for anaesthesia should be assessed for respiratory disease. It is useful to take a small blood sample for packed cell volume (PCV) assessment before anaesthesia when a prolonged procedure is anticipated.

BOX 10.1 Factors predisposing to anaesthetic problems in small birds

- Chronic malnutrition leading to immunosuppression, hepatic disease and cardiovascular disease
- Lack of exercise producing cardiovascular 'unfitness'

Hospitalisation facilities

Small kennels can be adapted to house birds, taking care to ensure escape is not possible. However, various sized cages are more appropriate for passerines and psittacines. Birds should always be hospitalised in a quiet area of the practice, as loud unfamiliar noises will cause stress.

Small birds may benefit from a hide box or nest compartment, for example a small cardboard box, during hospitalisation to reduce the stress of being in the unfamiliar environment. Nursing staff should still be able to observe the patient. If a pet bird is usually caged with or alongside a companion, it may be less stressful to hospitalise that bird with the patient.

Perches suitable for birds include wooden dowelling or non-toxic wood, such as apple tree twigs or branches. The size depends on the size of bird and provision of a variety of diameters will allow the bird to exercise its feet. Perches at different heights should be provided to allow the bird to move around within its temporary enclosure. Wooden dowelling is appropriate for most birds; non-toxic branches have the added advantage of providing environmental enrichment as the bird can chew them.

Parrots are less stressed if their hospital cage is in a high position, rather than placing the cage on the floor. However, they should not be allowed to perch above human shoulder height as this gives them a psychologically dominant position over staff.

The basal metabolic rate is higher in passerines than in non-passerines (Lasiewski and Dawson, 1967). Associated with this are rapid heat loss and utilisation of body energy reserves. Birds will, therefore, lose weight when not feeding, for example overnight, or if ambient temperatures are low (Perrins, 1979). To encourage birds to feed longer and reduce energy required for thermoregulation, hospitalised passerines should be provided with an extended day length (even 24 hours per day in some cases) and the environmental temperature raised (Coles, 1997).

Supplemental oxygen and heating

Ill birds benefit from supplemental heating during hospitalisation. Those with respiratory problems should also receive additional oxygen in an incubator or brooder (see Figs 9.5 and 9.6).

Fluid and nutritional support

Hypoglycaemia is common in malnourished or starved passerines, those with increased demands due to sepsis, or those with hepatic dysfunction. Paresis, depression and seizures may be seen. Treatment should be performed as soon as possible, causing as little stress to the patient as possible. Glucose solutions may be administered via crop-tube or given intravenously. If the patient is dehydrated, this should be addressed before dextrose is administered (Clippinger and Platt, 2000).

Puréed fruit and vegetables, or organic dairy-free fruit or vegetable baby food are suitable food substitutes for most species if crop feeding is required. Some birds, particularly hand-reared individuals, may voluntarily take egg food or hand-rearing formula from a spoon. Several enteral nutritional support formulas are available for ill birds, such as Harrison's® Recovery Formula (http://www.harrisonsbirdfoods.com/) or Critical Care Formula® (VetArk, Winchester, UK). These may be offered in a small bowl, but in general they are crop-fed to the patient throughout the day to ensure adequate intake of nutrients. In birds with strong beaks, such as the psittacines, a metal tube should be used for crop feeding or a metal speculum used to open the mouth to allow safe passage of a plastic tube.

The jugular vein is large and the most common site for phlebotomy or emergency access, but the superficial plantar metatarsal vein may be catheterised in quite small birds. A catheter can be readily stabilised in this latter vein for peri-anaesthetic fluid administration. Small gauge needles and catheters, usually 25–27 gauge, are used for venous access in these small patients to reduce the risk of venous damage.

Although intraosseous injections may be administered in very small birds, the risk of iatrogenic fracture precludes routine use in patients weighing less than 100 g. In birds weighing over 200 g, a 23-gauge, 25-mm hypodermic needle can be used for intraosseous access (Chitty, 2005).

Air sac cannulation is possible in small birds. For patients weighing 50–100 g, a dog or cat urinary catheter may be shortened, or a large-bore venous catheter used (Edling, 2005).

EQUIPMENT REQUIRED

Specialist equipment is not required for anaesthesia in these species. However, it is useful to have small endotracheal tubes for intubation during anaesthesia and various sizes of crop tube if nutritional support is required.

BOX 10.2 Basic equipment for small bird anaesthesia

- Digital scales for accurate weighing
- Small gauge needles and small syringes (e.g. insulin syringes)
- Towel for restraint during induction and recovery
- Supplemental heat source, e.g. heat pad or hot water bottle covered with towel
- Appropriate sized endotracheal tubes (or catheters with circuit attachments)
- T-piece circuit
- Monitoring equipment: stethoscope, echocardiogram
- Trained assistant to monitor patient while clinician performs procedures

BOX 10.3 More specialist equipment for small bird anaesthesia

- Mechanical ventilator
- Monitoring equipment: capnography, pulse oximeter, oesophageal echocardiogram
- Manufactured small endotracheal tubes

TECHNIQUES

Routes of administration

Injections

The jugular vein is commonly used for phlebotomy, and is large and accessible for emergency *venous access*. More commonly, an intravenous catheter is placed in the superficial ulnar vein (see Fig. 11.3) or superficial plantar metatarsal vein during anaesthesia. The superficial plantar metatarsal vein can be accessed even in psittacines with short legs (Fig. 10.2), but it can be difficult to insert a needle or thread a catheter proximally before the vein deviates; small gauge needles/catheters should be used to facilitate this. Needles and catheters are usually 23–25 gauge; catheters should be short. If an indwelling catheter is placed, care should be taken to position and secure the catheter to allow the bird to perch or move its wings comfortably. The catheter should, therefore, not extend below the foot and the bandage should not be cumbersome.

Intraosseous catheters can be placed in parrots with relative ease. In birds weighing between 200 g and 700 g, a 21-gauge, 25-mm hypodermic needle can be used (Chitty, 2005).

Intubation

Endotracheal tube placement may be more difficult in psittacines than in other avian species. The tongue is usually fleshy with the glottis located caudally at the base, and the strong beak means that the patient must be at a deep plane of anaesthesia to allow opening of the oral cavity without risking damage to either the clinician's fingers or the endotracheal tube (see Fig. 10.1). If volatile anaesthetic agents are used for induction via facemask, recovery will be rapid and there is little time to place the tube when the mask is removed. It is important to have all equipment ready to hand, including a length of tape for securing the tube once placed.

It is safer in larger species to use atraumatic forceps to grasp the tongue during intubation rather than placing your fingers into the mouth of lightly anaesthetised parrots, in order to avoid the patient biting on to your fingers if it recovers during the procedure.



Figure 10.2 • Intravenous catheter placed in the superficial plantar metatarsal vein of a male Eclectus parrot (*Eclectus roratus*).

PRE-ANAESTHETICS

Pre-medication is rarely used in these species. However, butorphanol has been shown to be anaesthetic-sparing when used in combination with isoflurane in cockatoos (Curro et al., 1994), and would be appropriate if analgesia is required.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

On anaesthetic induction, the oropharynx should be checked for the presence of ingesta, the crop assessed for the presence of food and the trachea intubated. If intubation is not possible, the neck should be raised above the level of the crop to reduce the risk of regurgitation and the beak lowered to allow drainage should any occur.

Injectable agents

The combination of ketamine with xylazine is not recommended in touracos (members of the order Cuculiformes, related to Passeriformes). Only low doses of ketamine and medetomidine are used in psittacines due to the questionable health status of most pet birds (Coles, 1997).

As a species widely available for research, many anaesthetic studies have been performed in pigeons. When induced with ketamine, medetomidine and butorphanol, arrhythmias were seen in some pigeons; as these ceased after atipamezole administration, the irregularities are likely due to medetomidine (Atalan et al., 2002).

Volatile agents

The minimum alveolar concentration (MAC) for isoflurane in cockatoos is 1.44% (Curro et al., 1994).

Anaesthetic maintenance

Most birds can be intubated, using either an endotracheal tube or a modified catheter. Anaesthesia is usually maintained with volatile agents via a mask or endotracheal tube.

Recovery

If volatile agents have been the sole anaesthetic, recovery is typically rapid. Most small birds are gently restrained in a towel during recovery. After prolonged procedures or with debilitated birds, it is useful to pre-warm an incubator or brooder for the post-anaesthetic period.

Suggested anaesthetic protocols

Small birds may be induced in a chamber to reduce the stress of handling, but it can be difficult to assess the depth of anaesthesia. When the righting reflex is lost, anaesthesia is maintained using either a facemask or intubation. Endotracheal intubation is possible in birds as small as 30 g, but for these patients the tube may need to be fashioned from an intravenous catheter (with the stylet removed) or soft rubber catheter. It is important to perform intermittent positive pressure ventilation (IPPV), as these small tubes are more prone to obstruction with airway secretions.

Anaesthesia is usually induced by administration of volatile agents via a facemask in larger birds (100 g and heavier). When applying a mask to a parrot's head, a swift motion should be used to pass the mask over the beak, as these species will tend to bite and damage the rubber or plastic of the mask. As the rim of the mask passes over the bird's head, take care not to damage the eyes.



Figure 10.3 • Indirect blood pressure measurement from the ulnar artery in a sulphur-crested cockatoo (*Cacatua galerita*).

ANAESTHESIA MONITORING

Observations on the patient

All birds should be closely observed throughout the anaesthetic period, monitoring respiratory movements, heart rate and reflexes.

Anaesthetic monitoring equipment

C*apnography* has been used to monitor isoflurane anaesthesia in African grey parrots (*Psittacus erithacus timmus*) receiving IPPV via a mechanical ventilator (Edling et al., 2001). $P_{ET}CO_2$ correlated well with $PaCO_2$, but overestimated it by approximately 5 mmHg in the birds.

Electrocardiogram (ECG) measurements have been performed in several species, including one study involving four species of macaws, genera *Anodorhynchus* and *Ara*, under isoflurane anaesthesia (Casares et al., 2000). A paper speed of 50 mm/s and calibration of 10 mm = 1 mV was used. Significant differences were found between species for heart rate and ECG measurements.

Blood pressure can be measured indirectly using an inflatable cuff and Doppler probe (Fig. 10.3). In patients weighing less than 300 g, the distal humerus is a more reliable site than the femur for cuff placement (Lichtenberger, 2005).

PERI-ANAESTHETIC SUPPORTIVE CARE

Fasting

Veterinary texts vary in their recommendations for fasting before anaesthesia. Malnourished or debilitated animals will benefit from provision of food as long as possible, but the crop should be allowed to empty before induction of anaesthesia.

Birds weighing less than 100 g should not be fasted before anaesthesia (Coles, 1997), as they are likely to become hypoglycaemic. The gastrointestinal transit time is fast in these small birds. If the patient is not eating, a crop-feed 1 h before anaesthesia will usually allow time for the crop to empty, and reduces the risk of hypoglycaemia. The crop should be checked on induction of anaesthesia and any ingesta present aspirated to reduce the risk of regurgitation and aspiration.

Larger birds may be fasted for a short period; usually crop emptying will occur in 2–3 h (Edling, 2006; Forbes and Altman, 1998). Birds in good condition larger than 100 g appear to cope with overnight fasting and water deprivation for 2–3 h without adverse effects (Franchetti and Kilde, 1978). In general, birds between 300 g and 1 kg are fasted for 6 h and those between 100 g and 300 g for 3–4 h (Coles, 1997).

Analgesia

Pain recognition can be particularly problematic in parrot species. They may vocalise excessively as normal behaviour, whilst concealing a true discomfort. Familiarity of the particular bird may allow the owner to detect subtle behavioural changes. Often the veterinary surgeon must rely on more obvious signs of pain, such as restlessness, inactivity or biting at a lesion to detect a problem.

Pigeons have been shown to have high proportions (76%) of kappa (κ) opioid receptors in their forebrain. Butorphanol may, therefore, be a better analgesic in pigeons than μ opioid agonists, such as morphine (Mansour et al., 1988).

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11 Birds of prey anaesthesia

INTRODUCTION

This chapter will discuss species from the orders Strigiformes (owls) and Falconiformes (vultures, hawks, eagles and falcons). Strigiformes contain two families, the Strigidae (most owl species [Fig. 11.1]) and Tytonidae (which includes the barn owl). Falconiformes are divided into five families, including the Falconidae (falcons) and Accipitridae (hawks, eagles and Old World vultures) (Perrins, 2004). Anatomical and physiological variations pertinent to anaesthesia of birds of prey will be described. Various anaesthetic protocols will be considered.

The owners of these species usually fly the birds regularly, either for pleasure or working (to collect vermin species), particularly Falconiform species. They are, therefore, often presented in good cardiovascular fitness, in contrast to most pet passerines and psittacines presented to the veterinary clinician.



Figure 11.1 • This female snowy owl (*Nyctea scandiaca*) belongs to the Strigidae owl family.

ANATOMY AND PHYSIOLOGY

Respiratory system

The most common cause of respiratory pathology in birds of prey is *Aspergillus* spp. (Redig, 1993). Infection presents most often as *Aspergillus fumigatus*. This fungus is an opportunistic organism and usually infects immunosuppressed birds (Dahlhausen, 2006). Predisposing factors include stress, confinement, poor husbandry, malnutrition, other disease, prolonged antibiotics or steroids (McMillian and Petrak, 1989; Redig, 2000). The spores are usually found in dusty, warm, humid environments with poor ventilation. Certain species are reported to be more susceptible to aspergillosis, including gyrfalcons (*Falco rusticolus*), goshawks (*Accipiter gentilis*), golden eagles (*Aquila chrysaetos*) and snowy owls (*Nyctea scandiaca*) (Forbes, 1991; Fransen and VanCulsem, 1988; Redig, 2000, 1993).

Urinary system

The nasal glands are functional in falcons, assisting with water homeostasis (O'Malley, 2005).

In cases of uricaemia, allopurinol may be used to decrease uric acid production, but renal toxicity has been reported in red-tailed hawks (*Buteo jamicensis*) receiving this drug (Lumeij et al., 1998). Carnivorous birds such as

birds of prey should be fasted before blood sampling, as false-positive elevations in blood uric acid levels will be seen in birds that have not been fasted (O'Malley, 2005).

Gastrointestinal system

Most species have a crop. Owls do not have an obvious crop, but rather a spindle-shaped distension of the cervical oesophagus (Taylor, 2000).

The diet for free-ranging birds of prey is predominantly small mammals and birds, although some species are adapted to hunt fish or other aquatic species. Nutritional imbalance is less commonly seen in captive carnivorous birds than frugivorous or granivorous species, as captive feeding of whole prey is similar to diets in the wild. Problems may be seen if one food item only is fed, rather than the variety that free-ranging birds would consume. For example, day-old chicks have a high fat content if the yolk-sac is not removed.

Birds of prey perform intestinal reflux or casting approximately 12 h after feeding, in order to void the indigestible parts of their diet, such as bone (in owls), fur and feathers (all species) (McLelland, 1979).

Although athletically fit, birds in flying condition are often maintained lean to encourage them to work well. If these birds become ill, hepatic glycogen reserves will deplete rapidly and hypoglycaemia may be present (Coles, 1997). In contrast, most falconers supplement their birds during the moult when they are not flying and these birds are less likely to be on the edge of nutritional imbalance.

Integumentary system

As many birds of prey have long tail feathers, they must be protected during hospitalisation to prevent damage that would hinder the bird's flight capabilities. A tail-guard (Fig. 11.2) made of paper, card or radiographic film should be placed around the tail to protect it, particularly if the bird is unable to perch.



Figure 11.2 • Tail-guard made from radiographic film on a Saker falcon (*Falco cherrug*).

Behaviour

Different species and individual birds react to hospitalisation and restraint in dissimilar ways. Some species are easier to handle, for example buzzards (*Buteo buteo*), while others are more aggressive and nervous, such as goshawks (*Accipiter gentiles*) or sparrow hawks (*Accipiter nisus*) (Coles, 1997).

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

Local variations and individual falconer preferences may affect the specific husbandry of birds presented to the veterinary surgeon. Details of enclosure design and components of the diet should be ascertained. The amount of food given is likely to vary seasonally, depending on whether the bird is flying, moulting or breeding. The source of food may be important, for example trichomoniasis infection from a diet of infected pigeons.

Most owned birds of prey will have restraint equipment and be trained to the fist, but should be cast with a towel to control the wings and legs in order to allow detailed clinical examination. Most species do not bite, but the talons of even small species can apply pressure and cause injury to the handler if not properly restrained.

Hospitalisation facilities

In contrast to other avian species, many raptors are accustomed to being tethered close to the ground. However, care should be taken to approach the bird at the same level, as an approach from above may be perceived as aggression.

Birds should be provided with a perch that is high enough to prevent the wing and tail feathers from trailing on the floor, where they may become damaged. Most trained birds of prey are kept tethered on a bow perch, with falcons more usually on a block perch. Perches are often covered with artificial turf (AstroTurf®, Southwest Recreational Industries, Texas), which spreads the load-bearing surface across the feet to reduce the risk of pododermatitis, and is easy to clean and disinfect. Alternatively, most birds will perch on a sturdy wooden branch during short-term hospitalisation. If the raptor is unable to perch high enough to protect its plumage, it is vital to protect the tail with a guard (see Fig. 11.2). The owner will not appreciate it if the bird's tail feathers become damaged during hospitalisation so the bird cannot fly until after a subsequent moult.

Most captive birds will eat from a selection of day-old chicks, mice, rats, rabbits and quail. It is important to ascertain what the bird's usual diet is, including quantities fed, before admittance to the veterinary practice. Although some birds will initially require easily digestible supportive nutrition, the aim will be to progress to the normal diet as soon as possible. The practice may store frozen food, such as day-old chicks, to defrost before feeding to

the patient. If the practice cannot source the food, the client can be asked to provide food for the bird during hospitalisation. Most raptors are fed once or twice daily.

Birds of prey should be fasted before elective procedures to allow for crop emptying. Feeding material that will not produce casting (i.e. meat without feathers and bones) will allow for feeding closer to the time of anaesthesia.

Fluid and nutritional support

Suitable nutritional support products for raptors include Hill's a/d® (Hills, Herts., UK), a convalescent food formulated for dogs and cats. In patients who have been anorexic for a period of time or where dehydration is suspected, fluids are provided first. Dehydrated patients may have gastrointestinal motility or absorption problems. In mild cases of dehydration, water or electrolyte solutions, such as Lectade® (Pfizer Limited, Sandwich, Kent, UK), should be administered orally before food is administered. If more severe dehydration is present, subcutaneous, intravenous or intraosseous fluids should be used to rehydrate the patient (in increasing order of effectiveness).

EQUIPMENT REQUIRED

Falconer's gloves and towels are necessary for restraint during induction. A variety of sizes of endotracheal tube should be available. Most birds of prey are sufficiently large to allow the anaesthetist to use multiple pieces of equipment to assist with monitoring of the patient, such as oesophageal stethoscope, electrocardiogram (ECG) and capnograph.

TECHNIQUES

Routes of administration

Injections

There are several sites for *intravenous* access in birds of prey. The jugular vein is ideal for phlebotomy and emergency administration of fluids or medication. The superficial ulnar or basilic vein is most commonly used for catheter placement peri-anaesthetically in patients over 300 g body weight (Fig. 11.3), although the superficial plantar metatarsal vein is another option. Needles and catheters used are 23–25 gauge, depending on the patient size. Indwelling catheters are secured using sutures to the skin or feathers and/or bandage materials. In most cases, the catheter is bunged and intermittent boluses of fluid or medication administered, but in some instances a continuous rate infusion will be administered. Raptors appear to traumatise catheters and drip lines less than other companion birds.

The circulation can also be accessed via the *intraosseous* route. In raptors weighing between 200 g and 700 g body weight, a needle similar to that used in parrots may be used for intraosseous access, i.e. a 21-gauge, 25-mm hypodermic needle. In birds weighing more than 700 g, an 18-gauge, 38-mm needle is more appropriate (Chitty, 2005).

Intubation

The tracheal opening is easily visible in most birds of prey. The tongue is typically long and thin, with the glottis at the base (Fig. 11.4). Birds of prey have relatively large tracheas, for example a 3–3.5 mm tube will be appropriate for a 750 g hawk, or a 4–5 mm tube for a 3 kg eagle.



Figure 11.3 • One electrocardiogram pad attaches to the skin on the ventral propatagium (with another on the other wing and the third over the thigh), and an intravenous catheter is present in the superficial ulnar vein (taped in place at elbow) of this Saker falcon (*Falco cherrug*) hybrid.



Figure 11.4 • Oral cavity of a common buzzard (*Buteo buteo*) showing the typical long thin tongue seen in Falconiformes with the glottis at the base.

PRE-ANAESTHETICS

A *benzodiazepine*, such as midazolam, may be administered intramuscularly to produce tranquillisation prior to mask induction with a volatile agent. The benzodiazepine will reduce the stress of induction, and the improved muscle relaxation is useful if surgery is to be performed. The effects of midazolam will persist for several hours (Valverde et al., 1990).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Injectable agents

Although *ketamine and a benzodiazepine* will produce deep sedation or anaesthesia with good muscle relaxation, recovery may be prolonged in raptors (Forbes, 1984). Ketamine is reported to produce poor-quality restraint and anaesthesia in raptor species (Ludders et al., 1989). However, ketamine has a wide safety margin and can be administered via any parenteral route, including orally. The addition of a benzodiazepine will reduce the risk of convulsions due to ketamine (Baronetzky-Mercier and Seidel, 1995).

If anaesthesia is induced with *ketamine and xylazine*, nocturnal raptors appear to metabolise the drugs more rapidly than diurnal species (Haigh, 1980). *Buteo* species appear to be sensitive to this combination, with deep anaesthesia and prolonged recovery times (Coles, 1984). In contrast, the goshawk (*Accipiter gentiles)* and Coopers hawk (*Accipiter cooperii*) required higher doses, but had associated prolonged recovery times (Redig, 1983). The combination of ketamine with xylazine is not recommended in large owls (Coles, 1997).

Another combination of a dissociative agent and benzodiazepine is the proprietary preparation of *tiletamine and zolazepam* (Zoletil®, Virbac Laboratories, Carros, France or Telazol®, Fort Dodge Laboratories Inc, Fort Dodge, IA). This preparation has been used to immobilise Eurasian buzzards (*Buteo buteo*) via parenteral injection (Trah, 1990) and up to 80 mg/kg orally (Zenker et al., 2000). Oral administration does not usually result in surgical anaesthesia, and the effects seen vary between powdered and liquid formulations. It has also been used successfully in several other species, at various doses that differ between species (Blyde, 1992; Gray, 1974; Hayes, 1996; Schobert, 1987).

Propofol has been used to induce and maintain anaesthesia via a continuous rate infusion in red-tailed hawks (*Buteo jamaicensis*) and great horned owls (*Bubo virginianus)* (Hawkins et al., 2003). Intravenous administration of propofol at 1 mg/kg/min induced anaesthesia. Parameters assessed during anaesthesia showed that the propofol had minimal effect on blood pressure, but effective ventilation was reduced. Recovery periods were prolonged, and central nervous system excitatory signs were observed.

Volatile agents

Anaesthesia is most commonly induced in raptors via a close-fitting facemask with a volatile anaesthetic agent, most commonly isoflurane. Initial high gas concentrations, for example 4–5% isoflurane, are used to induce anaesthesia. After induction, the bird is intubated and maintained on a lower concentration of anaesthetic, usually 1–3% isoflurane.

Anaesthetic maintenance

Birds are intubated and maintained on volatile agents. Intermittent positive pressure ventilation (IPPV) should be performed.

Recovery

Patients should be appropriately restrained during recovery, usually by casting in a towel. This is to reduce the risk of self-trauma during recovery and to avoid injury to staff. Most birds of prey do not fit in incubators and a large kennel in a warm room is used for recovery. A heat pad and towels or blankets can be used to warm and pad the kennel. The perch should be removed until the bird has recovered sufficiently to perch soundly.

Suggested anaesthetic protocols

As with other birds, isoflurane is generally used to induce anaesthesia via facemask for short procedures. Intubation will protect the airway and allow IPPV.

For more prolonged or painful procedures, injectable agents are administered before induction with isoflurane via a facemask to produce a more balanced anaesthesia. Midazolam is a useful pre-medicant in nervous or stressed birds, and opioids such as butorphanol will also provide some sedation as well as analgesia. Addition of these agents produces a smoother induction and recovery than volatile agents alone.

ANAESTHESIA MONITORING

Observations on the patient

In general, patient monitoring is similar to other anaesthetised birds. However, the toe pinch is unreliable as a method of assessing anaesthetic depth in birds of prey, as the response is variable depending on the patient (Coles, 1997). Respiratory rate, depth and pattern should be closely assessed, as should heart rate and rhythm.

Anaesthetic monitoring equipment

A bell or oesophageal *stethoscope* can be used to monitor heart rate and rhythm (Fig. 11.5).

ECGs (see Figs 9.8 and 9.13) have been recorded in various raptors, for example peregrine falcons (*Falco peregrinus*



Figure 11.5 • Intubated Bonelli's eagle (*Aquila fasciata*). In larger birds such as this, an oesophageal stethoscope can be used. The lead across the head is from the electrocardiogram machine.

brookei) (Rodriguez et al., 2004). However, values will vary in anaesthetised patients. One study has produced reference values for buzzards (*Buteo buteo*) anaesthetised with isoflurane (Espino et al., 2001). Generally ECG leads are attached as with other species, but adaptors can be obtained to obtain ECG measurements via an oesophageal probe (Cardio Companion ECG Probe and Esophageal Lead, SurgiVet Inc, Waukesha, WI).

Capnography has been used in several species of birds of prey (Desmarchelier et al., 2007). Arterial blood gas analysis was performed on samples from the superficial ulnar artery. This study reported a good correlation between arterial blood partial pressures of carbon dioxide and end-tidal measurements over a normo-carbic range. However, at low values of $P_{ET}CO_2$, the associated $PaCO_2$ was overestimated, possibly due to hyperventilation during the study. The $PaCO_2$ was underestimated at higher $P_{ET}CO_2$, possibly due to hypoventilation and dilution of the sampled gas with fresh gas from the anaesthetic circuit, along with a decrease in capnography accuracy at higher values of $P_{ET}CO_2$ (Edling et al., 2001; Teixeria Neto et al., 2002).

PERI-ANAESTHETIC SUPPORTIVE CARE

Fasting

Carnivorous species, such as birds of prey, tend to eat once or twice daily. In species with a crop, it will empty in 6–8 h (Forbes and Altman, 1998). These birds should be fasted for 12 h, to allow the previous meal to be digested and a cast produced. If the anaesthetic procedure is planned, it can be helpful to provide a feed without casting material (that is, meat without fur, feathers and bones) on the previous day. If a bird of prey is in poor body condition or dehydrated, it may be gavage-fed with a liquid food supplement or electrolyte solution up to a few hours before anaesthesia. This should allow time for the fluid to pass through the crop to the proventriculus.

Antifungals

Owing to the high susceptibility of birds of prey to aspergillosis, prophylactic anti-fungal agents, such as itraconazole, are routinely administered if the patient has any of the predisposing factors listed above. These will include stress associated with hospitalisation or other concurrent disease.

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12 Reptile anaesthesia

INTRODUCTION

Almost 8000 reptile species are divided into four orders: Testudines (Chelonia) (tortoises, turtles and terrapins), Crocodylia (alligators, crocodiles, gharial), Rhynchocephalia (tuatara) and Squamata (includes the suborders of snakes and lizards) (Halliday and Adler, 2004; Holz, 2006). Many species are endangered in the wild, and regional and global legislation may restrict their capture and movement. This chapter will cover those pets most commonly seen in veterinary practices, namely chelonia, snakes and lizards. Tuatara are unlikely to be seen as pets, but techniques are similar to those used in lizards. Crocodilians will be covered briefly, as smaller species owned by herpetologists may be presented to the veterinary practice.

Reptiles are considered an unusual pet, but are kept by a broad spectrum of people. Different species have different characteristics, with some being relatively easy to maintain in captivity and others only suitable for experienced herpetologists. Reptiles present to the veterinary clinician for a variety of reasons and case care may be for the single pet or for a group.

Anaesthesia may be required for a range of procedures in these species. Demeanour varies greatly across the orders, with some requiring little chemical restraint for procedures such as radiography, for example tortoises, while others require sedation for many procedures, such as snakes. Anaesthesia (with appropriate analgesia) is required for surgery in all species.

Contrary to older texts, more recent work suggests that reptiles do experience pain (Kanui and Hole, 1990, 1992). Techniques previously advocated for restraint of reptiles included chilling. This has been shown to slow the ectothermic animal's metabolism and responses, allowing easier manipulation, but not to provide analgesia. Hypothermia is painful and brain necrosis may also result (Schumacher and Yelen, 2006). Chilling is inhumane and not an ethically acceptable technique for restraint of reptiles. Many anaesthetic and analgesic agents have been used in reptiles. Although there are some challenges to anaesthetising these animals, many of these drugs are effective at providing immobilisation in a humane way.

> Chilling is not an ethically acceptable method for restraint of reptiles.

ANATOMY AND PHYSIOLOGY

Although basic anatomy is similar between species, significant variations are present, even within groups. This section will discuss general reptilian anatomy as it pertains to anaesthesia and peri-anaesthetic care, and aspects of husbandry that may predispose to disease with potential effects on anaesthesia. Later sections will provide more detail on different reptilian groups, although other texts should be consulted to obtain species-specific anatomy and physiology.

An appreciation of normal reptile anatomy and physiology will aid with pre-anaesthetic assessment of the patient's health and thereby suitability for anaesthesia. It will also provide the clinician with the knowledge to select appropriate anaesthetic agents, which may have undesirable side effects on the patient, and enable monitoring of the patient during anaesthesia and recovery.

Stress

Reptiles are particularly susceptible to captive husbandry inadequacies, frequently succumbing to disease when stressed (Cowan, 1980). Environmental conditions, especially temperature, are important for all body processes. As their metabolism tends to be slower than that of mammals or birds, and clinical signs are often difficult to identify, pathological processes are often not observed in reptiles until an advanced state. For this reason, it is important for veterinary practices to be able to provide appropriate hospitalisation facilities, and the clinician should endeavour to identify and correct problems that may affect post-anaesthetic survival.

As certain reptile species are dangerous, causing bites or scratches, or being venomous, staff safety is important when hospitalising these animals. All enclosures should be secured, to prevent animal escape leading to self-trauma or human casualties. Signs should be used to indicate venomous species and, if possible, enclosures should be locked. Latex or nitrile gloves should be worn when handling reptiles, to avoid transference of pathogens between patients or of zoonotic infections to handlers. Further personal protective equipment (PPE) may be required with some animals, for example the use of towels to restrain green iguanas or snake hooks with venomous snakes.

Temperature

Reptiles are unable to generate their own body heat, but rather they are ectothermic. (The two exceptions to this rule are the fat-laden, giant, leatherback sea turtle (*Dermochelys coriacea*) and the incubating female Indian python (*Python molorus*), which produce a small amount of heat (Bennett and Dawson, 1976; Seymour, 1982).) By depending on external environmental temperature for body heat, reptiles lower their metabolic requirements (Bennett and Nagy, 1977). However, low environmental temperatures limit locomotion and enzymatic activity in reptiles. Smaller animals are more susceptible to changes in the external temperature. Most reptiles are poorly insulated and readily lose heat (O'Malley, 2005a).

The reptile immune system is dependent on the maintenance of body temperature (Mitchell, 2006). At cooler temperatures immune responses are lower, with less antibody production and fewer circulating peripheral white blood cells (Mitchell, 2006; Rossi, 2006). Studies have shown that some reptiles self-regulate temperature when combating a pathogen; by selecting a higher environmental temperature, they will effect a 'fever response' to stimulate the immune system (Vaughn, 1963, 1974). Reptiles with advanced disease may not show this response (Mitchell, 2006).

Each reptile species has a preferred optimum temperature range (POTR), also called the preferred optimum temperature zone (POTZ). This is the natural temperature range in which free-ranging individuals will reside. Table 12.1 lists ranges for common pet species. Different metabolic functions will be optimal at different temperatures within this range (i.e. each has a preferred body temperature [PBT]) (Pough et al., 2002). It is important to maintain reptiles within their POTR during hospitalisation, otherwise metabolic processes, such as digestion, drug metabolism and healing, may function sub-optimally. This includes the anaesthetic period, when it will be necessary to work in a warm environment and provide supplemental heating for the patient. Heat lamps, electric heat pads, water bottles, forced air blankets and heated circulating water blankets can all be used to maintain the patient's body temperature during anaesthesia and the recovery period.

It is necessary to create a temperature gradient within a vivarium, so that the patient is able to select whether to warm up or cool down. For most diurnal species, a daytime background temperature range from 27°C to 35°C with a hotter basking area is appropriate; for nocturnal or montane species 21–27°C is advisable. Nighttime temperatures are lower, usually 21°C (Rossi, 2006). Most heat sources, for example bulbs and mats, can be connected to a thermostat. However, thermostats are not 100% reliable and it is wise to–monitor the temperature range, ideally using maximum–minimum digital thermometers (Fig. 12.1).

Most reptiles, including chelonia and diurnal lizards, derive heat by basking in the sun (heliothermy). Free-ranging nocturnal species may warm from contact with hot surfaces (thigmothermy). In captivity, it is usual to provide a basking heat source, for example a hanging heat bulb (Fig. 12.2) or a heat mat at the side of the vivarium. It is advisable to ensure animals cannot contact electrical heat sources, as thermostats are often unreliable and animals frequently obtain contact burns.

The cardiovascular system is involved in thermoregulation. The heart rate in reptiles increases in warmer temperatures, hastening body warming. In species with three-chambered hearts (see Cardiovascular section), the right to left cardiac shunt will also reduce cooling by avoiding evaporation from the lungs. Peripheral blood vessels are dilated during warming and then constricted to reduce cooling (Pough et al., 1998b). Lymphatic vessels are large in reptiles and attempted venous access commonly accesses lymph.

Other husbandry factors

Poor husbandry will affect the patient's health in reptiles more than in any other exotic pet. Many species are maintained in countries with a climate far removed from their native land and, as ectotherms, rely very much more on their owner for provision of an appropriate external environment, including supplemental heating for many species. Table 12.1 lists some species commonly seen, with basic husbandry requirements that should be adhered to when patients are hospitalised.

Several problems may be seen in animals maintained in inappropriate environments. Outwith the species' POTR, the body's enzymes will function sub-normally. An inappropriate environmental temperature will adversely affect all metabolic processes in the body. Immunosuppression may occur, leading to an increased susceptibility to infections. Inappropriate temperature and relative humidity often lead to dysecdysis (which may require surgery, for example retained spectacles in snakes and some geckos). As with mammals, insufficient (or excess) ventilation will predispose the animal to respiratory tract infections. Inappropriate diets in reptiles often contain insufficient or unbalanced quantities of certain minerals. Ultraviolet (UV) lighting is required for many species (particularly herbivores) to allow calcium metabolism. Diets may also contain excess fats (for example, insectivores fed primarily on grubs).

UV light is necessary for many reptiles, with most chelonia and lizards having an absolute requirement. UV-A (320–400 nm) affects behaviour and appetite, while UV-B

Table 12.1: Husbandry and physiological information for common pet species (conscious values)

SPECIES	PREDOMINANT DIET	POTR (°C)	RELATIVE HUMIDITY (%)	COMMENT
African spurred tortoise (*Geochelone sulcata*)[2]	Herbivorous	25–35	40–75	Don't hibernate. Diet mainly grasses/hay
Asian water dragon (*Physignathus concincinus*)[4]	Herbivorous	24–30	80–90	Arboreal, semi-aquatic
Bell's hingeback tortoise (*Kinixys belliana*)[2]	Omnivorous	24–28	50–80	May hibernate in the wild, but usually over wintered in captivity
Boa constrictor (*Boa constrictor*)[4]	Carnivorous	28–30	50–80	–
Box tortoise (*Terrapene carolina* spp.)[1,2]	Carnivorous-omnivorous (depending on species)	21–27 (night >15, basking 27–32)	95	Hibernate
Burmese python (*Python molurus*)[4]	Carnivorous	25–30	50–80	–
Corn snake (*Elaphe guttata*)[4]	Carnivorous	25–30	30–70	–
Desert tortoise (*Gopherus agassizii*)[2,3]	Herbivorous	20–32	<30	Hibernate
Garter snake (*Thamnophis* spp.)[4]	Carnivorous	21–28	50–80	Diet mainly fish
Green iguana (*Iguana iguana*)[4]	Herbivorous	25–35	75–100	Arboreal
Inland bearded dragon (*Pogona vitticeps*)[4]	Omnivorous	25–35, basking area 38–42	30–40	Appreciate climbing facilities
Kingsnake spp. (*Lampropeltis* spp.)[4]	Carnivorous	25–30	30–70	–
Leopard gecko (*Eublepharis macularius*)[4]	Insectivorous	25–30	30–40	–
Leopard tortoise (*Geochelone pardalis*)[2]	Herbivorous	25–35	40–75	May hibernate in the wild, but often over-wintered in captivity
Map turtle (*Graptemys* sp.)[2]	Omnivorous	21–28 (water)	–	Some species hibernate

(*Continued*)

Table 12.1: (Continued)

SPECIES	PREDOMINANT DIET	POTR (°C)	RELATIVE HUMIDITY (%)	COMMENT
Marginated tortoise (*Testudo marginata*), North African tortoise (*T. graeca*), Hermann's tortoise (*T. hermanni*)[2]	Herbivorous	20–28	30–50	Hibernate
Red-eared slider (*Trachemys scripta elegans*)[2]	Omnivorous	20–24	60–90	Semi-aquatic. Young quite carnivorous, become more herbivorous as mature. Hibernate
Red-footed tortoise (*Geochelone carbonaria*)[2]	Herbivorous	21–27	50–60	Diet mainly fallen fruits, leaves and flowers occasional carrion. Do not hibernate
Royal python (*Python reguis*)[4]	Carnivorous	25–30	50–80	–
Savannah monitor (*Varanus exanthematicus*)[4]	Insectivorous	26–38	20–50	–
Spiny-tailed lizard (*Uromastyx* spp.)[4]	Herbivorous	20–25	50–90	–
Veiled (Yemen) chameleon (*Chamaeleo calyptratus*)[4]	Insectivorous	21–38	75–80	Arboreal. Require water via drip system or misting
Yellow-footed tortoise (*Geochelone denticulata*)[2]	Herbivorous	25–27	75–80	Does not hibernate

Key: POTR = preferred optimum temperature range

[1] (Boyer and Boyer, 2006); [2] (Highfield, 1996); [3] (McArthur et al., 2004a); [4] (Varga, 2004)

(290–320 nm) is necessary for vitamin D_3 manufacture and calcium absorption (Rossi, 2006). As UV light cannot pass through glass, any reptiles not kept outdoors in natural sunlight require UV supplementation. This is usually provided in the form of a UV strip light or bulb.

Relative humidity requirements vary with species, with some requiring high humidity and others comparatively dry environments. In the latter, a small area with high humidity is required, for example a box with damp substrate, particularly during ecdysis. Each species has specific water requirements (see Urinary section). Attention to conditions in the wild and provision of microclimates within a captive enclosure are important for many species.

As with other captive animals, ventilation of the enclosure should be considered. This can be difficult for animals requiring high environmental temperatures, or small species where escape may be possible through large vents. In non-climbing species, including most chelonia, a portion of the top of the enclosure can be open or covered with mesh to allow airflow. In smaller or more agile species, the use of wire netting is required over openings in the vivarium. Increasing ventilation should not allow the environmental temperature to drop below the POTR, and should not allow draughts to pass through the enclosure. Animals with respiratory disease that may be airborne should be maintained in a separate airspace to other patients. Feeding, cleaning and treatment utensils should also be disinfected between animals to prevent transfer of pathogens.

Where vivaria are not available, dog or cat cages can be modified. With small animals, a small Perspex box can be placed within the cage to retain the patient. Supplemental heating and lighting can be provided just outside or within



Figure 12.1 • Digital maximum–minimum thermometers can be used to monitor temperature in vivaria.



Figure 12.2 • Ceramic bulbs used to provide heat in vivaria should be covered with wire mesh to prevent patient contact resulting in burns.

the enclosure. Environmental parameters, for example temperature, should continue to be recorded. Due to the increased ventilation with these enclosures, more heating may be required than for a vivarium.

Most animals benefit from a hide in their enclosure. This may be a simple cardboard box or a log hide, or a plastic box with moist substrate (damp kitchen roll or sphagnum moss) for animals undergoing ecdysis. Arboreal species should be provided with branches for climbing. If the front of an enclosure is glass, most species benefit from an opaque strip at the bottom to prevent self-trauma during escape attempts.

An animal's response to various husbandry factors, such as inappropriate environmental temperatures or the presence of an aggressive companion, may result in a stress response. This response results in physiological changes that attempt to allow the individual to adapt to its environment. If stressors are severe or persistent, for example if enclosure temperatures are excessively high or low, the animal's physiology may not be able to adapt. In these situations, one outcome may be inhibition of the immune system, leading to an increased susceptibility to infections. Certain disease processes, such as chronic endoparasitism, may reduce the reptile's ability to respond to other disease and concurrent pathologies will be more likely.

Although most species will have specific substrate requirements for long-term care, most can be maintained on newspaper during hospitalisation. This allows for ease of vivarium cleaning. Water should be provided for all reptiles to drink. A deeper bowl may be required for soaking, but a shallow bowl should be provided for weak animals that may drown. Supervised soaking in deeper water may be appropriate during the day to encourage water intake (see Fluid therapy section below). Aquatic or semi-aquatic species obviously also require water in which to submerse.

BOX 12.1 Record-keeping

- Record previous and current captive husbandry in case notes
- Monitor and record husbandry during hospitalisation: vivarium temperature range, faecal/urine/urate production, appetite, medications received (including route), and treatments performed

Metabolism

Reptiles are ectothermic, relying on their environment for heat. As noted above, rates of metabolic processes within reptiles will thus be affected by external temperature. In general, the metabolic rate is much slower in reptiles compared to similar sized mammals (Bennett and Dawson, 1976). Each species will have a specific metabolic rate (Bennett, 1972; Espinoza and Tracy, 1997), with an optimum

temperature for this metabolism within their POTR. Smaller species or those that actively hunt have higher metabolic rates to cope with a high metabolic demand. Larger species may increase their metabolic rate after feeding to facilitate digestion (Secor and Diamond, 1995; Secor and Nagy, 1994).

When bursts of activity are required, reptiles can convert to anaerobic metabolism for short periods (Bennett and Licht, 1972). Lactate build-up causes an acidaemia, with an associated reduction in haemoglobin oxygen affinity, delaying oxygen transport (Pough et al., 1998a). Right to left cardiac shunting occurs during this process (Machin, 2001).

Few studies have investigated drug efficacy in reptiles. Likewise, little is known of drug absorption, distribution, metabolism and excretion in these species. Most reports of drug use are sporadic and in small numbers of animals. Due to the large variation in physiology between reptile species, there are likely to be differences in drug effects that preclude direct extrapolation between species, even those within the same taxonomic order. Illnesses, such as renal or hepatic dysfunction, or hypoproteinaemia associated with parasitism or nutritional deficiencies, are also likely to affect drug metabolism. Dehydration and hypothermia have profound effects on drug metabolism, and many reptiles require correction of these parameters before administration of other medications (Mitchell, 2006). Clients should be advised before their pet is admitted that data are limited and that most drugs are not specifically licensed for use in reptiles.

Cardiovascular system

Reptile hearts are located within the cranial part of the coelomic cavity. In chelonia, the heart lies midline, cranial to the liver and just dorsal to the plastron. In most lizard species, the heart is at the level of the pectoral girdle, although in larger species, such as monitors (*Varanus* sp.), it lies more caudally. The serpentine heart lies within the cranial body third (Funk, 2006).

The heart in snakes, lizards and chelonia is three-chambered, with two atria and one ventricle (Fig. 12.3). A pressure differential exists between the chambers, ensuring that oxygenated and preoxygenated blood do not mix (White, 1976). Changes in pulmonary resistance will allow shunting of blood either towards (during respiratory activity) or away from (during oxygen starvation, for example diving or apnoea) the lungs. Use of the right to left shunt allows the lungs to be bypassed, reducing oxygen loss from the circulation and also reducing evaporative heat loss. A pathological increase in pulmonary resistance, for instance pneumonia, would similarly shunt blood away from the lungs. This shunt becomes important when using volatile agents, as uptake of the anaesthetic into the systemic circulation will be reduced.

Crocodilians have a heart similar to mammals, with two atria and two ventricular chambers.

Ventilation and apnoea will affect blood flow through the pulmonary and systemic circulation in reptiles (Kik and Mitchell, 2005). Research with sliders (*Trachemys scripta* spp.) has shown that the shunt between pulmonary and systemic circulations is mediated by adrenergic mechanisms,

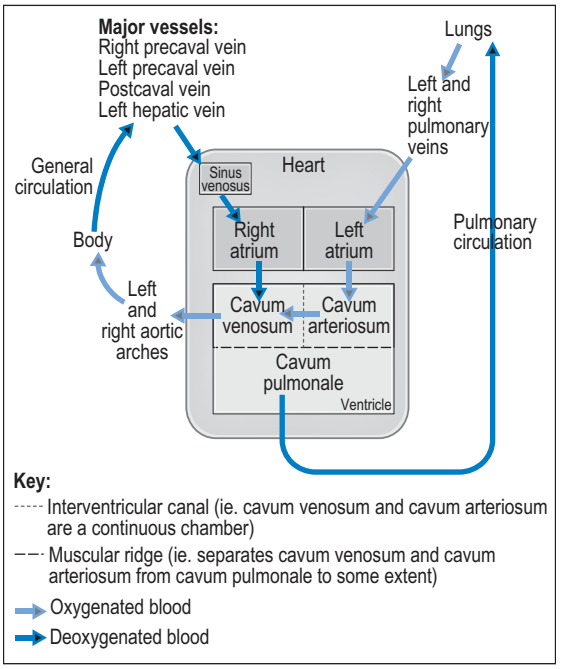


Figure 12.3 • Schematic showing circulation in non-crocodilian reptiles.

BOX 12.2 Circulation in reptiles (non-crocodilian)

- Heart = right atrium (with sinus venosus), left atrium and ventricle (comprising cavum venosum, cavum arteriosum and cavum pulmonale)
- Cavum venosum and cavum arteriosum form a chamber, with the interventricular canal in between
- Left and right single-cusped atrio-ventricular valves arise from the interventricular canal and prevent regurgitation from the ventricle to the atria
- A muscular ridge within the ventricle separates the cavum venosum and cavum arteriosum from the cavum pulmonale
- Timing of muscular contractions and pressure variations in the heart creates a functionally dual (systemic and pulmonary) circulation system during normal respiration. For example, in normal respiration in a red-eared slider (*Trachemys scripta elegans*), 60% of the cardiac output is directed to the pulmonary circulation with 40% to the systemic circulation; the pulmonary circulation is bypassed during diving (Williams, 1992)
- When pulmonary resistance is increased, for example during diving or in animals with lung pathology, a right to left shunt occurs. Blood is, therefore, recycled around the systemic circulation and bypasses the pulmonary circulation

but blood flow is primarily determined by the ratios of resistance within the two circulations (Overgaard et al., 2002).

The normal blood volume in reptiles is approximately 5–8% of body weight (O'Malley, 2005a). A 100 g snake would, therefore, have 5–8 ml of blood. Reptile haemoglobin is different to that in mammals and there are also species differences. In reptiles, haemoglobin tends to have a lower oxygen affinity, allowing release of oxygen to tissues at very low blood oxygen levels. During hypoxic conditions such as diving, particularly during exercise where production of lactic acid leads to metabolic acidosis, the Bohr effect again increases the release of oxygen from blood to tissues (Murray, 2006a). The oxygen-carrying capacity of a reptile's blood is greatest within the species' POTR (Davies, 1981).

Heart rate in reptiles increases with temperature, as does peripheral vasodilation, leading to increased peripheral perfusion and thus increasing heat intake. The opposite occurs with reduced environmental temperatures, conserving heat. In order to maintain cardiac output at lower temperatures, stroke volume is increased (White, 1976). Heart rate is increased during periods of activity, often threefold (Murray, 2006a). Smaller animals tend to have a higher heart rate than larger individuals. Other factors that will elevate heart rate include increases in metabolic rate, respiratory rate and sensory stimulation (Davies, 1981).

The renal portal system (see Urinary section) allows blood to drain from the pelvic limbs into the kidney (O'Malley, 2005b). This may have some bearing on administration of medications into the caudal portion of the reptilian body, but it is thought that valvular control may allow this to be bypassed at certain times (O'Malley, 2005a).

BOX 12.3 Reptile cardiovascular system

- Snake, lizard, and chelonian hearts are 3-chambered
- Crocodilian hearts are 4-chambered
- Reptiles are capable of anaerobic metabolism
- Environmental temperature will affect heart rate and perfusion

Respiratory system

Reptiles generally breathe through their nostrils. The nasal passages are connected via the internal nares to the oral cavity and thence the glottis. The anatomy of the airway will vary with species, but the glottis is generally quite rostral. Snakes protrude the glottis and trachea out of the mouth to allow simultaneous breathing and feeding (O'Malley, 2005a).

There are two arytenoid cartilages and one cricoid cartilage surrounding the glottis. The glottal opening is closed at rest, opening during inspiration and expiration. It is found on the base of the oral cavity, but the position varies in different species. In snakes, the glottis is quite rostral on the tongue (see Fig. 14.9). Glottal position in lizards depends on the species, being rostral in most species, but more caudal in herbivores (see Fig. 13.3). The chelonian glottis is at the base of the fleshy tongue (see Fig. 15.1), while the crocodilian glottis is behind the epiglottal flap. Crocodilians have several features that allow underwater hunting and breathing at the water's surface with their mouth open underwater, including the presence of this epiglottal flap (also known as the basihyoid plate or valve) at the base of the tongue (see Fig. 16.1) (Diaz-Figueroa and Mitchell, 2006).

Snakes and lizards have incomplete tracheal rings, but chelonia and crocodilia have complete tracheal rings (Davies, 1981). The trachea bifurcates at the level of the thoracic inlet in chelonia, at the level of the heart in snakes, and at the base of the heart in lizards.

The mucociliary apparatus is primitive in reptiles, predisposing them to inhaled infections and reducing their capabilities of clearing exudates from the airways (Murray, 1996). Although reptilian lung volumes are large, they have a much smaller surface area (approximately 1%) than similar-sized mammals (Wood and Lenfant, 1976). The lungs of reptiles are essentially sac-like, with the surface area increased by various invaginations, which form faveoli (equivalent to mammalian alveoli) lined with respiratory epithelium.

Serpentine bronchi are short, leading into elongated, sac-like lungs. Boids have a vestigial, but functional, left lung, while other species have a right lung only. Lizard lungs are single-chambered (unicameral) in most species, but multi-chambered in iguanids. Iguana and chameleon lungs are paucicameral, having a few chambers, but no intrapulmonary bronchus. Snake and some lizard lungs extend into air sacs (see Fig. 13.5) with non-respiratory epithelium. Chelonia have left and right intra-pulmonary bronchi, leading to paired lungs that are multi-chambered (multicameral) and relatively rigid. Monitor lizards also have multicameral lungs. Crocodilian bronchi branch into multiple internal lobes, with complex multi-chambered lungs (Perry, 1989; Perry and Duncker, 1978). Reptilian lungs are delicate, being easy to inflate due to high compliance (Schumacher and Yelen, 2006). Over-inflation and rupture during positive pressure ventilation (PPV) are also easy.

The respiratory epithelium varies between reptile species. In all species, gas exchange occurs across the falveolar epithelium of the lungs, but less so in the caudal lungs of snakes. No respiration occurs in air sacs. Other accessory respiratory surfaces include the skin in some soft shell aquatic species, buccopharyngeal mucosa in many lizards and cloacal mucosa in terrapins (Seymour, 1982; Wood and Lenfant, 1976). Softshelled turtles (*Apalone* spp.) are able to obtain up to 70% of their oxygen requirements through their shell when underwater (Marcus, 1981). Cutaneous sites of respiration are also important for carbon dioxide elimination in aquatic species (Schumacher and Yelen, 2006).

Reptiles do not possess a diaphragm as found in mammals. Crocodilians are the exception, with a muscular septum similar to the mammalian diaphragm caudal to the lungs. In order to breathe, intrapulmonary pressures are changed by musculature movements, including the intercostals, pectoral and abdominal muscles. Chelonians have a non-muscular pseudo-diaphragm (the septum horizontale), which separates the lungs from the rest of the coelomic cavity. Limb movements stretch this septum, pulling it ventrally and creating negative pressure, and, thus, inspiration (Davies, 1981).

Respiratory control varies between species. In snakes it is primarily centrally controlled. There are species differences in the presence or absence of intrapulmonary chemoreceptors and stretch receptors; many snakes have both while many chelonian species have stretch receptors only (Redrobe, 2004).

The respiratory rate of reptiles varies with environmental temperature and body size, but is usually 10–20 breaths per minute (O'Malley, 2005a). In a normal breathing cycle, a reptile will expire, inspire and relax. This last phase can be particularly long in aquatic animals (Wood and Lenfant, 1976). Reptiles tolerate hypoxia well, converting to anaerobic metabolism during periods of apnoea (Murray, 1996). Their myocardium tolerates this change in metabolism better than mammalian myocardium, with species variability in this tolerance (White, 1976). Heart rate reduces during apnoea and the right-to-left cardiac shunt predominates, bypassing the pulmonary circulation (Murray, 2006a).

Respiration is related to oxygen and carbon dioxide concentrations, and environmental temperature. The main stimulus to breathe is low levels of oxygen in blood, causing an increase in respiratory rate. High levels of carbon dioxide will lead to an increase in tidal volume. An elevated temperature, or a prolonged dive in aquatic species, will lead to elevated oxygen demand that is met by an increase in tidal volume (not an increase in respiratory rate). High levels of environmental oxygen lead to reduced ventilation, due to a decrease in respiratory rate and tidal volume (Wood and Lenfant, 1976).

Reptiles are able to function well with anaerobic metabolism. Circulatory buffering systems enable the animals to tolerate acid–base imbalances produced by lactic acid and hydrogen ion build-up during anaerobic periods. Consequently, reptiles are able to cope during hypoxia produced by respiratory illness, masking clinical signs until pathology is severe and the individual is unable to compensate further (Murray, 1996). A poor ability to move inflammatory exudates from the lungs will further compromise the reptile with respiratory pathology, as the increase in pulmonary resistance will tend to direct cardiac blood flow towards the systemic circulation.

BOX 12.4 Reptile respiratory system

- Chelonia and crocodiles have complete cartilaginous tracheal rings
- Most snakes only possess a right lung (boids have a vestigial left lung also)
- Low partial pressure of oxygen is the main stimulus to breathe
- Respiratory pathology results in:
  - an increase in pulmonary resistance, leading to right-to-left shunting of blood in the heart and reduced supply to the pulmonary circulation
  - an increase in anaerobic metabolism to compensate for hypoxia

Several factors will predispose to disease in the reptilian respiratory system. These include stressors discussed earlier, such as malnutrition, ecto- or endoparasitism, excessively high or low relative humidity, poor hygiene or suboptimal environmental temperature (Murray, 2006b). Bacterial pneumonia is common in reptiles, either as primary or opportunistic pathogens. Most infections are associated with aerobic Gram-negative isolates, such as *Aeromonas*, *Klebsiella*, *Proteus*, *Pseudomonas* and *Salmonella* (Hilf et al., 19990). Viral, parasitic, and fungal infections are also reported. High environmental humidity, low temperature and dusty substrates may increase fungal spores in the environment.

Rhabdias nematodes are found in snake and lizard lungs, and pentastomids may be throughout the respiratory tract in all species (Murray, 2006b). Various trematodes may infect the respiratory tract (Murray, 2006b) and other nematodes may have a pulmonary larval migrans (Stoakes, 1992). Occasionally foreign bodies may be inhaled. Other disease processes, such as space-occupying lesions, may also compromise respiratory function.

Owing to the respiratory depressive effects of anaesthetic agents, it is prudent to stabilise the patient with respiratory disease prior to anaesthesia. In most species many investigations can be performed without anaesthesia, including radiography and tracheal swabs for cytology and/or culture. If oxygen supplementation is deemed necessary, care should be taken not to depress respiratory drive further in reptiles by elevating inspired oxygen levels.

Urinary system

In common with other vertebrates, water constitutes approximately 75% of a reptile's body weight (66% in chelonia) (Fig. 12.4) (Minnich, 1979; Smits and Kozubowski, 1968). Cellular and endothelial membranes between water compartments are selectively permeable. Intracellular ion concentrations produce osmotic forces to maintain water,

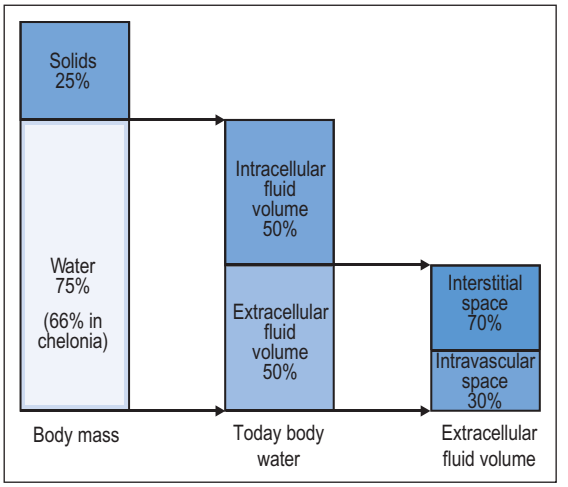


Figure 12.4 • Water distribution in reptiles.

Starling's forces act at vascular membranes to maintain water and plasma proteins act as impermeable solutes to produce colloid osmotic pressure within the compartments (Mader and Rudloff, 2006).

Water homeostasis in reptiles differs significantly from that in mammals. The slow reptilian metabolism results in slower synthesis of water. Reptiles are unable to produce more water than their evaporative water loss and so are more likely to become dehydrated when not drinking or absorbing water via other methods (see Fluid therapy section) (Minnich, 1979). Reptilian kidneys are much less well developed than those of mammals and birds, with fewer and shorter nephrons, fewer capillaries to the glomeruli, and no loop of Henle (Dantzler, 1976; Holz, 2006). Kidneys are responsible for synthesis of vitamin C (Gillespie, 1980) and active vitamin D (Illrey and Bernard, 1999).

The paired kidneys are connected to the urinary bladder or cloaca via ureters. Not all reptiles have a bladder, for example agamidae lizards have only rudimentary bladders. Snakes, crocodilians and some varanidae lizards have no bladder (Fox, 1977). Owing to their lack of renal reabsorption of water, homeostasis in reptiles relies on their drinking water, reabsorbing it from the cloaca and bladder, and salt loss via the nasal salt glands (Fitzsimmons and Kaufman, 1977).

Reducing the glomerular filtration rate also conserves water. If a reptile becomes dehydrated, blood may cease to flow through the glomerulus. There is a dual blood supply to the reptilian kidney, from the renal arteries and renal portal vein (similar to avian species, see Fig. 9.2). The renal portal vein bypasses the renal glomerulus, entering the tubules directly. This mechanism ensures renal perfusion continues even when glomerular blood flow is reduced to conserve water (Murray, 2006a), preventing ischaemic necrosis of the renal tubules. A valve for regulating the amount of blood entering the kidneys via the renal portal system has been described in Red-eared sliders (*Trachemys scripta elegans*) (Holz et al., 1997b), which may be affected by adrenaline (epinephrine) and acetylcholine as in poultry (Rennick and Gandia, 1954). Shunts also bypass the kidneys by carrying blood from the renal portal system to the postcava (Porter, 1972). Historically, texts have advised against administration of drugs in the caudal half of the reptilian body, but more recent work suggests that the site of administration does not lead to nephrotoxicity or subtherapeutic levels (Holz et al., 1997a; Holz et al., 2002).

Most species will drink water and so should be provided with a shallow bowl of water when hospitalised. A deeper bowl or aquarium is necessary for semi-aquatic or aquatic species. Chameleons require dripping water (Fig. 12.5). After anaesthesia, waterbowls should be removed until the patient has recovered sufficiently to ensure drowning does not occur. Aquatic animals should be intermittently sprayed with water during recovery to avoid integument desiccation.

As mentioned above, the reptile urinary system differs significantly from that of mammals. Most mammals excrete waste products from protein and amino acid metabolism as urea in water. As reptiles cannot concentrate urine, water must be conserved. Terrestrial species excrete nitrogenous waste products as insoluble uric acid, which forms the white or yellow urates seen in reptile excreta. Aquatic reptiles excrete ammonia, urea and small amounts of uric acid.

Reptiles will also regulate water loss by selecting a certain environmental humidity, and attention should be paid to species' humidity requirements during hospitalisation. Water is reabsorbed from the urinary bladder (found in chelonia and some lizard species) or (in species without a bladder) refluxed from the urodeum into the rectum where protein, electrolytes and water can be reabsorbed. Water can also be absorbed from the cloaca (Braun, 1998; Dantzler, 1976; Schmidt-Nielsen and Skadhauge, 1967). This mechanism can be utilised for rehydrating reptile patients, as they may absorb water if soaked in a shallow warm water bath.

Lipids on the skin reduce water loss in reptiles. Skin permeability also increases when in contact with water, allowing transdermal rehydration (Lillywhite and Maderson, 1982). Under-hydrated reptiles may become anuric and urates may form solid concretions. Alternatively, uric acid may build up in the body and lead to gout. Many reptiles excrete potassium and sodium via an extra-renal salt gland to conserve water (O'Malley, 2005a).

BOX 12.5 Reptilian urinary tract is adapted for water conservation

- Few nephrons
- Low glomerular filtration rate
- Renal portal system
- Uric acid production (terrestrial species)

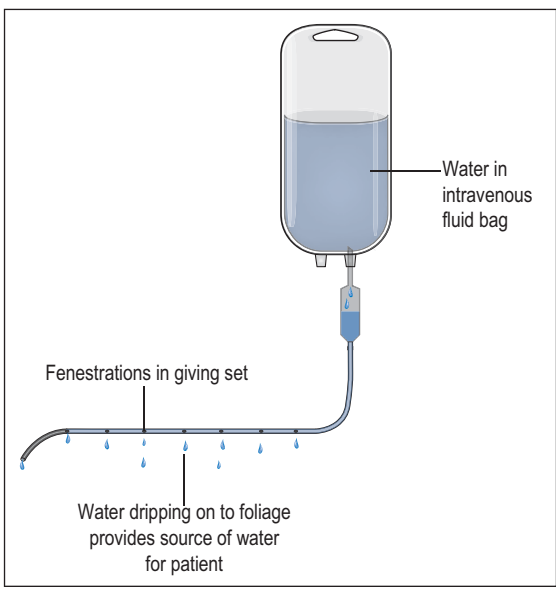


Figure 12.5 • Simple set-up for administration of water in a chameleon enclosure. (A plastic bottle with pinholes in the base could also be used to provide water.)

Renal diseases have been reported in many species of reptile. Pathologies include inflammatory or degenerative changes, infection or neoplasia. Metabolic derangements may lead to amyloidosis or gout (Frye, 1992, 1994a). Diets containing high levels of protein predispose to renal disease in herbivorous reptiles (Barnard, 1996; Jacobson, 2003). Chronic dehydration will also affect renal circulation and thereby function (Hernandez-Divers and Innis, 2006). Clinical signs associated with renal disease are often non-specific and further investigation (blood and urine analysis, radiography, ultrasound and renal biopsy) is necessary to assess fully for renal pathology and obtain a prognosis.

Digestive system

Reptiles fall into three broad categories according to their diet: herbivores, carnivores and omnivores (Table 12.1). Each species will have specific requirements and it is advisable to maintain a captive diet as close to that in free-ranging conspecifics as possible. The reader is advised to consult other texts for specific dietary information.

The clinician should ascertain the patient's nutritional status. Inappropriate diets or insufficient calorific intake will cause the reptile to become dehydrated, have muscle wasting and necrosis, and compromised hepatic (usually associated with lipidosis) and renal function (Mitchell, 2006). Malnourished animals in a state of negative energy balance may be hypokalaemic and hypophosphataemic, and susceptible to refeeding syndrome with associated cardiac dysfunction (Donoghue and Langenberg, 1996). During hospitalisation, many animals, therefore, require supportive care, particularly to stabilise debilitated animals before anaesthesia (Tables 12.2, 12.3 and 12.4). The reptilian oesophagus is quite fragile (Diaz-Figueroa and Mitchell, 2006) and gavage feeding should be performed with care using sufficient lubrication.

The digestive tract in reptiles varies depending on the species' diet (see Table 12.1), with herbivorous species having quite complicated tracts compared to carnivores (O'Malley, 2005a). The positioning of organs within the body cavity also varies with species, with major differences found in the elongated body of snakes. All reptiles have a single combined excretory–reproductive organ, the cloaca (Diaz-Figueroa and Mitchell, 2006).

The oral mucosa should be moist, but colours can vary from pale pink to grey (Diaz-Figueroa and Mitchell, 2006), with some species being highly pigmented. Chelonians do not have teeth. Those in other species vary, for example being long and caudal-pointing in snakes. Acrodont teeth,

Table 12.2: Fluid rates for reptile rehydration (Mitchell, 2006)

Maintenance requirements = 1–3% of total body weight; i.e. 10–30 ml/kg/day In addition, replace fluid deficits over 72–96 h
Oral fluids: volume administered <2–3% body weight per dose

Table 12.3: Fluids commonly used in reptiles, with their distribution characteristics

Crystalloids
Replacement fluids (avoid lactated solutions if hepatic disease present): Lactated Ringer's solution – ECF (interstitial and intravascular) 0.9% saline – ECF (interstitial and intravascular) 7% saline – ECF (intravascular) 5% dextrose in water – ICF Maintenance fluids: 2.5% Dextrose in half-strength lactated Ringer's solution – ECF (interstitial and intravascular) Jarchow's solution (two parts 2.5% dextrose in 0.45% saline, one part lactated Ringer's solution) – ECF (interstitial and intravascular)
Colloids
Hydroxyethyl starches, for example Hetastarch – ECF (intravascular), synthetic Whole blood – ECF (intravascular), natural

Key: ECF = extracellular fluid compartment; ICF = intracellular fluid compartment

(Mader and Rudloff, 2006)

Table 12.4: Suggested foods for supportive feeding via gavage. Blended normal diet or commercial enteral diets are often more balanced than baby foods

DIETARY GROUP	FREE-RANGING/ CAPTIVE	SUPPORTIVE FEEDING IN DEBILITATED ANIMALS
Herbivore	Leaves, fruits, flowers and vegetables	Herbivore support diet (e.g. Oxbow® Critical Care for Herbivores), puréed fruits or vegetables, organic and dairy-free vegetarian baby food
Carnivore	Small mammals (rodents), frogs, insects	Dog/cat support diet (e.g. Hills® a/d), blended killed prey
Omnivore	Molluscs, small mammals, fruits, vegetables	Combination of the above, puréed killed prey (e.g. insects), organic and dairy-free baby food

for example in bearded dragons (*Pogona vitticeps*) are not replaced (Diaz-Figueroa and Mitchell, 2006). The clinician should be aware of species' peculiarities during restraint, particularly during oral manipulations of venomous species.

As with other metabolism in reptiles, the rate of digestion is related to body temperature and thence to environmental temperature. Hospitalised animals should be maintained at their POTR to allow normal digestion, particularly if recently fed. Dietary composition and species-specific gastrointestinal anatomy will also affect gastrointestinal transit time (King, 1996). These factors impact on anaesthesia as reptiles may regurgitate ingesta, particularly if recently fed or maintained at too low an environmental temperature.

Gastrointestinal transit time is usually 2–4 days in small carnivorous lizards and snakes. Passage in large snakes, herbivorous chelonians and lizards may take up to 3–5 weeks (Diaz-Figueroa and Mitchell, 2006). Fasting is advisable before anaesthesia of carnivorous species to reduce the risk of regurigitation, either during anaesthesia or as a response to restraint.

Nervous system

The spinal cord extends to the tip of the tail in reptiles (Bennett, 1996a). There is no true subarachnoid space, and the subdural space lies between the leptomeninges and dura mater (O'Malley, 2005a).

Many veterinary clinicians do not provide analgesia for reptiles. However, all vertebrate species experience pain. These pain pathways have not been fully elucidated in reptiles. Reptiles have been shown to possess an endogenous opioid system and primary Aδ-nociceptive neurons are described in snakes (Liang and Terashima, 1993; Ng et al., 1986).

As with many other exotic species, it can be difficult to assess reptiles for the presence of pain. Reptiles do not vocalise as some other species do and behavioural changes are likely to be subtle. An appreciation of the species' and, preferably, individual animal's normal behaviour may allow the clinician to identify changes resulting from pain. This is more difficult if the species presented is unfamiliar to the clinician or if a chronic illness has resulted in insidious onset of clinical signs that the owner has not noticed. Often the animal will be less active and spend more time hiding, but some may be more aggressive. An elevated respiratory or heart rate may accompany pain. The reptile may 'guard' the painful anatomical body part, for example non-weight-bearing lameness with limb pain or an arched back with coelomic discomfort.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

Many reptiles presented for anaesthesia will have either subclinical or overt disease, which may be chronic in nature, but have an acute onset of clinical signs. Often animals have been anorexic for long periods, resulting in poor body condition. Inappropriate diet or husbandry may result in severe dehydration. Debilitated animals often succumb to bacterial and fungal infections. The aim of the pre-anaesthetic period is to identify any problems and treat them if possible, or manage them to reduce the risks associated with anaesthesia. As discussed above, inappropriate environments or diets often cause or predispose to disease in captive reptiles; for this reason, a thorough review of current and previous husbandry should be ascertained to identify adverse factors.

If husbandry factors suggest sub-clinical disease may be present or clinical signs of disease are seen, further investigation should be undertaken to identify the extent of pathology and its aetiology. Many reptiles are not presented to the veterinary clinician until disease processes are advanced. These patients are often debilitated and, thus, are very poor candidates for anaesthesia. It is advisable to provide supportive care prior to anaesthesia in these animals, possibly providing fluid and nutritional support for several days if procedures requiring anaesthesia can be postponed while the patient is stabilised. If anaesthesia cannot be postponed, a more guarded prognosis should be given and peri-anaesthetic supportive care, for example fluid therapy, should be more aggressive.

> It is important to provide an appropriate environment for hospitalised reptiles before and after anaesthesia. Maintenance of the patient within the species' POTR is vital and the environmental temperature should be monitored using a digital thermometer.

History

This will include current and previous husbandry details for the patient. It is particularly important with these ectothermic species to note the temperature range in which they have been maintained. Any deviation from the species-specific POTR is likely to affect metabolic processes, including the immune system, and predispose to pathology that may affect anaesthesia. Any previous illnesses may also be pertinent to the enquiry. An appreciation of the animal's current appetite will enable assessment post anaesthesia.

A risk factor for infectious diseases includes contact, either directly or indirectly, with other reptiles. The owner should be questioned regarding other pets as well as indirect contact with other reptile collections.

Clinical examination

Observation of the patient before handling gives the clinician an opportunity to assess mobility, including the animal's resting stance. Weak animals are often inactive and quadrupedal animals may not be able to raise themselves above the ground in normal posture. Body condition can sometimes be assessed visually by observing muscle mass over bony prominences, such as vertebral spinous processes or pelvic bones, or fat reserves in the tails of species, such as leopard geckos (*Eublepharis macularius*). Respiratory rate and pattern are also best observed before restraining the patient. The skin of many species, particularly lizards, changes colour when unwell, often becoming darker. Ocular,

nasal or oral discharges may be seen. Species such as leopard geckos store fat in their tails and loss of a normal plump tail-base is suggestive of chronic malnutrition or illness.

A full clinical examination should be performed, to identify any current illness. (This may be restricted in dangerous or venomous species.) Respiratory assessment will include measurement of rate, depth and character of breathing. Normal reptilian respiration should be inaudible, but respiratory abnormalities can often be heard by placing your ear close to the animal in a quiet room. Although auscultation is possible using a stethoscope in snakes, lizards and crocodilians, it is often insensitive in detecting respiratory lesions (Chitty, 2004). A damp towel placed between the stethoscope and scales or carapace may aid auscultation. Mucous membranes should be assessed and any nasal or oral lesions noted that might pertain to respiratory disease. Doppler blood flow monitors are useful for cardiac assessment, placing the probe directly over the heart to obtain a heart rate, rhythm and character (Fig. 12.6).

The external body surface should be examined for swellings, lesions or ectoparasites. The abdominal region can be palpated in lizards, but this is a less sensitive technique for detecting abnormalities in snakes. Palpation and ballottement in the prefemoral fossae of chelonia may identify fluid or masses such as eggs.

Dehydration can be difficult to detect in reptiles; signs may include sunken eyes (particularly in chelonia, which suggests dehydration more than 8–10%), viscous oral mucus, reduced urine output, dry faeces, dysecdysis, presence of dry loose skin folds, lethargy and anorexia (Donoghue, 2006). A basic neurological examination can be performed. The eyes and ears should also be examined.

The body should be palpated to assess body condition, and an accurate body weight should be obtained. A comparison between weight and body length (snout to cloaca in snakes and lizards, or sub-carapacial length in chelonia) is also useful to assess body condition. These assessments enable calculation of drug doses, fluid and nutritional requirements.



Figure 12.6 • Doppler monitor used to monitor heart rate in a sedated African spurred tortoise (*Geochelone sulcata*).

Investigation

Further tests may be required to characterise any problems found on history and clinical examination. It is important to maintain the patient at a sufficiently warm temperature during these investigations. As with animals undergoing anaesthesia, supplemental heating can be provided from a number of sources (see Chapter 1).

Faecal analysis should be routinely performed in reptiles, particularly wild-caught animals that often have high endoparasite burdens. Parasitism may lead to debilitation and predispose to further disease in the individual. Samples of faeces or discharges may be necessary for culture, and should be taken prior to instigation of anti-microbial therapy.

Clinical examination and blood analysis will allow determination of the level of dehydration (Schumacher, 2000) Blood analysis will provide data on metabolism. As a minimum database, haematocrit packed cell volume (PCV), total protein and glucose should be recorded. The total blood volume in reptiles is 5–8% of body weight, and up to 10% of this can be safely taken from a healthy animal. Thus, a 500 g animal would have a blood volume of 25–40 ml, and 2.5–4.0 ml could be sampled. However, many reptiles are not healthy and a smaller volume is usually taken. A blood sample may not be possible pre-anaesthesia in many small patients; in these cases more reliance is based on clinical assessment. As ethylene diamine tetraacetic acid (EDTA) may lyse reptilian red blood cells, a fresh blood smear is used to estimate total and differential white cell counts. Lithium heparin samples can be used for biochemical tests.

Blood sample volume

The maximum blood sample size that should be taken from a healthy reptile is 0.8% of body weight, i.e. 0.8 ml from a 100 g animal.

Imaging may be useful to assess for internal pathology. Radiography and ultrasonography may be indicated, and can usually be performed in conscious reptiles. Where masses are present, cytology and microbiology may be performed on aspirates or biopsies. Anaesthesia will be required for certain procedures, such as lung washes and endoscopic investigations. Computed tomography (CT) and magnetic resonance imaging (MRI) techniques also require anaesthesia to immobilise the patient for prolonged periods.

Stabilisation

This stage of reptile anaesthesia is often the most critical. Many patients are presented with chronic debilitating disease and failure to stabilise prior to administration of anaesthetic agents may have fatal consequences. Initial triage should allow a decision to be made as to how long anaesthesia may be delayed, including any procedures for which the anaesthesia is required, and balanced against the benefits of stabilisation and supportive care.

Provision of an appropriate environmental temperature is vital in reptile medicine. Studies have shown that maintaining the reptile's body temperature at or just above the species POTR will be beneficial. The immune response is stimulated, with improved leukocyte function and reduced healing time. Fluids and medications will also be better absorbed and metabolised in the warm patient (Mader and Rudloff, 2006).

Most reptiles will be dehydrated and fluid therapy is vitally important. The simplest method of administering fluids is by immersion in a warm water bath or electrolyte solution, at the species' PBT. Weak animals should be continuously monitored to prevent accidental drowning.

In some patients, particularly those that have been anorexic, it may be necessary to provide nutritional support. Rehydration therapy should initially be administered to avoid 're-feeding syndrome' in hypophosphataemic and hypokalaemic animals (Bounos, 1972; DaSilva and Migliorini, 1990). Calorific requirements should also be gradually introduced, with 50% on the first day, 75% on the second and 100% on the third day (Calvert, 2004).

Fluid therapy

Dehydration is common in reptile patients, due to several factors. Inappropriate husbandry and diet or illness may lead to reduced water intake; excess losses may occur due to haemorrhage, reflux, vomiting or diarrhoea. History, clinical examination and blood results will allow an estimation of the patient's dehydration and fluid requirements. These parameters are reassessed following fluid administration. Heart rate will increase in the hypovolaemic reptile, providing it is within its POTR. Normal PCV is 20–40% in most reptiles and total solids (measured on a refractometer) 40–80 mg/L. Increases in both of these suggest dehydration due to reduced water intake or increased loss; decreases suggest acute haemorrhage has led to water depletion. Serum sodium and chloride levels will also be increased in dehydration (values are species-dependent) (Mitchell, 2006).

Oral fluid therapy can be used in patients for maintenance requirements or to replace water deficits if less than 5% dehydrated, provided the gastrointestinal tract is functional. This is usually via oesophageal or stomach tube, although small volumes may be administered via a syringe directly into the oral cavity (Mitchell, 2006). When more than 5% dehydration is present, parenteral fluids should also be administered.

The subcutaneous route is not used regularly in reptiles. The poor blood supply means absorption is slow, and the small subcutaneous space limits the volume that may be administered. This route should again only be used for reptiles less than 5–6% dehydrated (Mitchell, 2006).

Patients with moderate-to-severe dehydration can be given intracoelomic fluids. As with all fluids, these should be warmed prior to administration. There is a risk of organ puncture with this technique, but large volumes can be administered and absorption is rapid via the coelomic membrane and serosal surfaces. Fluid volume should be limited as reptiles lack a diaphragm.

Intravenous or intraosseous fluid administration is advisable for replacement of intravascular deficits. Sites for venous access are discussed in the species subsections, with most requiring surgical cut-down to access. Continuous rate infusion of intraosseous fluids is easiest or small boluses can be repeated.

> Warm fluids before administration to avoid inducing hypothermia.

Interstitial dehydration occurs when dehydration is more than 8% and may be clinically evident as tacky mucous membranes and sunken eyes. Intracellular hydration can be estimated after restoration of perfusion and interstitial hydration (Mader and Rudloff, 2006). Free water deficits should be replaced over 12–24 h (see Table 12.2). Dehydration present for more than 48 h should be replaced more gradually, over 24–48 h.

The choice of fluids for rehydration will depend on where water is required within the body. Isotonic crystalloids are often used to restore tissue perfusion, but the addition of colloids will reduce the volume required. Isotonic crystalloids used for fluid replacement (see Table 12.3) should have sodium levels similar to normal plasma to produce less of an osmotic gradient, while those for maintenance usually have lower levels. Fluids should be warmed to 30–35°C before administration (Mader and Rudloff, 2006).

The crystalloid 5% dextrose in water is hypotonic and will thus increase intracellular fluid deficits, distributing evenly across the total body water compartments. Hypertonic saline (7%) will increase the intravascular osmotic pressure, drawing fluid from the interstitial and intracellular compartments. This crystalloid should not be used in reptiles that cannot tolerate rapid increases in intravascular volume, for example those with dehydration, hypernatraemia, hyperchloraemia or hyperosmolality. Isotonic crystalloids, for example half-strength lactated Ringer's solution and 2.5% dextrose, will distribute across the extracellular fluid volume, with 70% entering the interstitial space and 30% remaining intravascularly.

Intravascular volume depletion is treated with isotonic crystalloids, usually lactated Ringer's solution or Jarchow's solution (see Table 12.3). Buffered fluids may correct acidaemia if present, but some studies suggest the buffer may exacerbate hyperlactataemia (Prezant and Jarchow, 1997). Reptiles can tolerate high levels of lactate, as produced during anaerobic metabolism (White, 1976). End-stage liver failure or liver hypoperfusion may allow the lactate salts in lactated Ringer's solution to affect plasma levels (Lowery et al., 1971). In cases of hyperkalaemia, hypercalcaemia or hypochloraemic metabolic alkalosis, 0.9% sodium chloride solution can be used for fluid therapy (Mader and Rudloff, 2006).

Excess isotonic crystalloids may reduce intravascular colloid osmotic pressure or further increase hydrostatic pressure, leading to movement of fluid into the interstitial compartment. This may compress lymphatic vessels and reduce drainage. Oxygen perfusion may be reduced if

anaemia is present, particularly if cardio-respiratory disease is also present. Colloids should be given in these cases (Mader and Rudloff, 2006).

Colloid fluids are isotonic, but contain large molecules that will remain in the intravascular fluid space and thus increase osmotic pressure. They must be administered intravenously or intraosseously. Colloids will, therefore, only increase intravascular fluid volume, but will do so for longer periods than the use of crystalloids alone. Concomitant administration of crystalloids and colloids will prevent interstitial dehydration, which may result if colloids are used alone. Commonly used synthetic colloids include hydroxyethyl starches and dextrans.

Whole blood transfusions are a form of colloid. The donor animal should be of the same species and healthy, and blood volume collected should be no more than 0.8% of body weight. Little is known about blood transfusions in reptiles, but in animals that have received transfusions previously a slide agglutination test can be performed (Mader and Rudloff, 2006).

BOX 12.6 Pre-anaesthetic stabilisation

- Failure to stabilise a patient prior to anaesthesia may have fatal consequences
- Fluid therapy should be administered to all patients before anaesthesia

Nutritional support

Reptiles that have a reduced appetite may respond to force-feeding, for example animals with reduced vision may eat if food is placed near or into the mouth. However, many reptiles presented to the veterinary clinician are completely anorexic and require intensive supportive care. Fluid deficits should be addressed before nutritional support, as the gastrointestinal tract will function sub-optimally in dehydrated patients.

Assist-feeding may be carried out using gavage feeding or, for patients likely to require longer-term support, via indwelling feeding tubes. Initial foods may be more liquid to assist with fluid losses, progressing to higher calorie foods for debilitated animals and eventually foods close to the normal diet for maintenance.

Feeding tubes come in a variety of types and sizes. Commercial tubes can be used for most patients and intravenous catheters (without the stylet) used for small patients. Gags or speculums are often required, as soft tubes should be protected from the patient's bite and the patient's jaws should be protected from damage by metal tubes. The volume of food administered and the feeding frequency depend on the size and health status of the reptile. Smaller animals will benefit from more frequent feeds to mimic their natural behaviour, but this must be balanced against a need to minimise stress to the patient.

EQUIPMENT REQUIRED

Anaesthetic equipment

Various sizes of *facemasks* and *induction chambers* may be of use in reptile anaesthesia. *Endotracheal tubes* come in a variety of sizes, with adaptations from intravenous and urinary catheters necessary for many small reptile species (see Fig. 1.6).

Since most reptile pets are less than 10 kg in weight, a non-rebreathing anaesthetic *circuit* should be used. For small patients (<10 kg), a T-piece is appropriate. The minimum gas flow rate is 1 L/min, as the vaporiser is inaccurate at lower flow rates. Larger patients may require other circuits (see Chapter 1).

Anaesthetic monitoring equipment

An 8 MHz *Doppler* probe is useful for assessing heart rate. Oesophageal *stethoscopes* may be used in larger animals. *Electrocardiography* (ECG) can be used in reptiles. Pulse oximetry is of limited use in reptiles and blood pressure monitors are rarely used.

Hospitalisation requirements

Additional *heat sources* are required during anaesthesia and the peri-anaesthetic period. These may include heat lamps, electric heat pads, water bottles, forced air blankets and heated circulating water blankets. The environmental temperature can be increased using central heating in treatment rooms, and heat mats and bulbs in vivaria. Anaesthetic recovery time will be prolonged in an environment outside the individual species' POTR, as metabolic processes including drug metabolism will be slowed.

Equipment for *fluid administration* in reptiles may include a sink or cat litter tray to use as a bath, oesophageal tubes (dosing catheters, urinary catheters), oesophagostomy tubes (Fig. 12.7), intraosseous needles or catheters



Figure 12.7 • Oesophagostomy feeding tube in a Hermann's tortoise (*Testudo hermanni*).

BOX 12.7 Basic equipment required to hospitalise and anaesthetise reptiles

- Secure enclosure – vivarium or kennel
- Easy to clean substrate, e.g. newspaper for terrestrial animal
- Heat source for enclosure and for anaesthesia, e.g. ceramic heater, heat bulb, heat mat or combination
- Digital thermometer to measure body and environmental temperatures
- Ultraviolet lighting (not required for all species)
- Water provision – varies from shallow bowl for some species to drink (Fig. 12.8), to aquatic tank for others to swim
- Hygrometer to monitor relative humidity
- Hide, e.g. cardboard box, upended broken plant pot
- Climbing facilities for arboreal species – e.g. wooden branch for bearded dragon
- Food – varies between species and whether self-feeding or assist-feeding. Ask owner to provide normal diet if practice does not normally stock
- Restraint devices – towels, gloves, snake hooks
- Ventilator or Ambu bag (self-inflating bag-valve-mask)
- Endotracheal tubes – 1–4 mm, uncuffed
- Oral gags – metal, wooden, plastic, tape; can be improvised (e.g. wooden tongue depressor)
- Small syringes and needles (insulin syringes) for small patients
- Infusion pumps or syringe-drivers for accurate fluid administration

to use in place, syringe-drivers or infusion pumps. Various fluids should also be on hand for use as required.

TECHNIQUES

Routes of administration

Most animals will absorb *oral* fluids well; these are usually administered in boluses via a rubber or metal oesophageal tube. A variety of tubes are available for this purpose. If a metal dosing catheter with ball-tip is used, a gag should be placed in the mouth to prevent trauma to the reptile's jaw, particularly in lizards and snakes where teeth may be damaged. If a rubber or polyvinyl chloride (PVC) dosing catheter is used, the gag is necessary to prevent the patient biting through the tube. A wooden tongue depressor covered with adhesive bandage is useful as a mouth gag in lizards. A smooth round plastic or wooden bar, for example a cotton-bud, can be used in snakes. For small or medium-sized terrestrial chelonia the clinician's finger can be placed in the corner of the mouth, while a wooden or metal gag may be more useful in larger or more aggressive chelonian species.

Pre-measure the length of tubing to reach the distal oesophagus or stomach. This will be approximately half-way down the length of the body in most reptiles. Prime the tube with fluids, to avoid introduction of relatively large volumes of air. Lubricate along the tube, for example with a small amount of petroleum jelly or sterile lubricating jelly, particularly in dehydrated patients where the delicate oesophageal mucosa may easily be traumatised. With the mouth opened, the glottis should be visualised. The tube for gavage feeding is passed dorso-laterally to avoid entering the glottis, which is closed at rest in reptiles. In chronically debilitated animals, a prolonged recovery may be anticipated and

BOX 12.8 Drug/fluid administration

- Prepare the skin aseptically before injections in reptiles, to reduce the risk of introducing infection
- In some species scales are present to provide protection, and the needle should be inserted between scales in these animals to avoid damaging the integument
- Pre-warm any fluids or food to be administered, to help maintain the patient's body temperature. Also ensure that fluids are not over-heated as this could cause internal damage



Figure 12.8 • Cat litter trays can be used to bathe reptiles to encourage drinking and voiding of waste products (such as this Hermann's tortoise [*Testudo hermanni*]), or as the water source for aquatic species within the hospital vivarium.

an oesophagostomy feeding tube deemed appropriate; the reader is referred to other texts for description of this technique (McArthur et al., 2004a; Mitchell, 2006).

Where more severe dehydration is present, or suspected, more aggressive fluid therapy should be instigated. Subcutaneous or intracoelomic fluids are not well absorbed in cold patients. The needle is inserted between scales at a shallow angle for *subcutaneous* injections. For *intracoelomic* injections, the animal is turned into lateral recumbency, to allow organs to fall away from the injection site in an attempt to avoid accidental organ puncture. The caudal third of the coelomic cavity is entered, to avoid the lungs or air sac (see Fig. 15.4D).

Constant rate infusions administered intravenously or intraosseously are more beneficial in debilitated animals, where intravascular fluid deficits are likely to be present. The intravenous or intraosseous routes are more difficult to access in reptiles, but in debilitated patients are the methods of choice for fluid administration.

A cut-down technique is necessary for placement of *intravenous* catheters in reptiles. In all but the most severely debilitated patient, anaesthesia or analgesia is required. Local anaesthesia is preferred to general anaesthesia for this procedure, as fluids are usually required to stabilise the patient to reduce the risks of general anaesthesia. Peripheral venoconstriction is common in dehydrated and hypothermic animals, and warming the animal before catheterisation will assist with peripheral venodilation (Mader and Rudloff, 2006).

Intraosseous access is used for patients that are either too small or with a circulation too collapsed to allow venous access. Intraosseous injections should be performed using appropriate analgesia, usually under general anaesthesia. (The intraosseous route is not possible in snakes.) The tibial crest is accessible in most species. The stifle is flexed, local anaesthetic injected and a needle with stylet inserted into the tibial plateau. This should avoid the joint capsule. The distal femur can also be used, entering the anterior surface. Correct positioning can be checked using radiography. Tape or sutures are used to secure the needle.

Intubation

Intubation is particularly important in anaesthetised reptiles as the relaxed position of the glottis is closed. Endotracheal intubation, therefore, maintains the trachea in an open position during anaesthesia, permitting passage of gases. Connecting the tube to an anaesthetic circuit also protects the airway from fluids, for example haemorrhage during oral procedures, and enables positive pressure ventilation (PPV) to be performed.

Adequate sedation or anaesthesia is required to produce jaw relaxation for intubation. (The exception is snakes, where conscious intubation is sometimes performed.) A good light source is necessary for the procedure. In species such as chelonia or herbivorous lizards, where the glottis is located caudally in the mouth, a laryngoscope is useful to aid visualisation.

Endotracheal tubes come in a wide variety of shapes and sizes. Uncuffed tubes are usually used, to avoid damage to the delicate tracheal lining that may result in ischaemic necrosis. The endotracheal tube should be as large as possible, to reduce escape of anaesthetic gases into the working environment, and to allow sufficient airway ventilation. If a cuffed tube is used, for example during oral procedures in larger animals, care should be taken not to over-inflate. Alternatives to cuffed endotracheal tubes are those with a step or 'shoulder' to place at the glottis, producing a wider base and narrow tip (see Fig. 1.6).

For very small animals, intravenous catheters or feeding tubes can be attached to connectors for anaesthetic circuits. The clinician should attempt to minimise functional dead space within the circuit by ensuring the extent of endotracheal tube between the patient's oral cavity and anaesthetic circuit is minimised (see Fig. 13.2). PPV is important in these patients to reduce the risk of respiratory secretions obstructing the endotracheal tube.

Assisted ventilation

Bradypnoea or apnoea occurs during anaesthesia in reptiles and they thus require assisted ventilation. This can be performed either by an assistant or by using a mechanical ventilator. Manual ventilation is labour-intensive and excess pressure can easily result in damage to the delicate airways. Mechanical ventilation is preferred, both because management is easier and also because the pressure or volume and respiratory frequency can be electronically maintained (Schumacher and Yelen, 2006).

The principles of intermittent positive pressure ventilation (IPPV) are described in the introductory chapter. The tidal volume in reptiles is much larger than that in mammals, but with a lower respiratory rate. Peak airway pressure should be less than 10–15 cm H_2O. A point particularly worth noting for manual ventilation is that inspiration should proceed over a maximum of 1 or 2 s. IPPV is usually performed at four to eight breaths per minute (Schumacher and Yelen, 2006). The reptile's body should be observed during forced inspirations and movements should mimic those seen during conscious inspirations in healthy animals. There is a tendency to over-inflate the lungs during forced ventilation. A low pressure should be used in reptiles as their have very fragile lungs and air sacs.

During mechanical ventilation, the pressure required to approximate normal inhalation will vary between individual animals. As a guide, this will be 5–10 cm H_2O in lizard species. In snakes, 6–10 cm H_2O is usually sufficient, but a higher pressure setting may be required to achieve full ventilation in the long trachea. In chelonia, 8–10 cm H_2O should increase intra-coelomic pressure and produce small outward movements of the limbs as seen in normal respiration.

Staff safety with venomous species

Many snake species and two lizard species (the helodermes) are venomous. Staff safety is paramount if these species are to be treated at a veterinary practice. If staff are not sufficiently trained or confident, it may be advisable to refer these animals to a better equipped practice rather than risk a potentially fatal incident. In general,

access to the oral cavity should be restricted to anaesthetised animals and great care should be taken not to self-envenomate when examining the oral cavity or intubating the animal. Emergency protocols should be in place for instances where a member of staff is bitten or the animal escapes.

BOX 12.9 Assisted ventilation

- Reptiles require assisted ventilation during anaesthesia
- This can be performed either manually or mechanically
- Peak airway pressure $<10–15$ cm H_2O
- Ventilation rate 4–8 breaths/minute

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Volatile agents

Gaseous anaesthetic agents are ideal for maintenance of anaesthesia in reptiles. These agents have a wide safety margin and recovery is rapid. In animals that do not breath hold, these agents can also be used for rapid induction of anaesthesia. In other species, including chelonia, injectable agents (see below) must be used to induce anaesthesia before inhalational agents can be instigated.

Isoflurane causes minimal cardio-respiratory depression, and also limited hepatic and renal toxicity. In unpre-medicated animals, anaesthesia can be induced in a chamber or via a mask with 5% isoflurane. Once the patient is sufficiently anaesthetised, as noted by loss of jaw and muscle tone, the animal is intubated (if possible) and maintained on 2–3% isoflurane (Schumacher and Yelen, 2006). Animals that are debilitated or have received pre-medication with other anaesthetic agents will require lower concentrations of inspired anaesthetic agent.

Sevoflurane has been used in reptiles with varying success. Concentrations of 7–8% were required for induction, with 3.5–4.5% to maintain surgical anaesthesia. Anaesthesia was not achieved in some species (Schumacher and Yelen, 2006).

Other routes of administration

Oral administration of sedative or anaesthetic agents in reptiles leads to unreliable results. Similarly, prolonged and unreliable induction times result from *subcutaneous* drug administration (Schumacher and Yelen, 2006). Therefore, anaesthetic agents are usually administered *intramuscularly*, *intravenously* or *intraosseously*. Drug pharmacokinetics do not appear to be significantly altered if anaesthetic agents are administered caudally, despite historical concerns regarding the renal portal system (Benson and Forrest, 1999; Holz et al., 1997a).

As in other species, the use of high doses of single agents results in pronounced cardio-respiratory depression and prolonged recovery times. The use of combinations of agents allows the clinician to reduce individual drug doses and thereby reduce side effects. This also allows the clinician to address analgesia and muscle relaxation of the patient.

Injectable agents

Ketamine hydrochloride is commonly used as part of the reptile anaesthetic protocol. Its advantages include the possibility to administer intramuscularly or intravenously, and a wide safety margin. However, muscle relaxation is poor and analgesia minimal. Recovery can be prolonged, particularly when used at high doses. Combinations of ketamine with benzodiazepines and opioids result in smooth induction and recovery, and provide muscle relaxation and analgesia (Schumacher and Yelen, 2006). Another dissociative anaesthetic, *tiletamine*, has been used in reptiles as the preparation containing *zolazepam*. Higher doses may have extended recovery times of up to 72 h.

Medetomidine is usually combined with ketamine and an opioid agent, for example butorphanol. This combination can be used to allow examination and minor procedures on larger species. Medetomidine and ketamine can be administered intramuscularly or intravenously. *Atipamezole* can be used to reverse the alpha-2-agonist.

Propofol is the anaesthetic agent of choice in reptiles. The safety margin is wide, induction is smooth, and recovery is smooth and rapid. The major disadvantage is the requirement for intravenous or intraosseous access for administration of this agent. Some animals may require pre-medication with a low dose of the agents discussed above before intravenous access is possible. Doses required vary between species, and in general snakes require a slightly lower dose and chelonia a higher dose compared to lizards (Divers, 1996). A continuous rate infusion or intermittent administration of boluses can be used to maintain anaesthesia (Schumacher and Yelen, 2006). As in other species, cardio-respiratory depression occurs with propofol. A rapid bolus injection may lead to apnoea (Bennett et al., 1998). The dose is, therefore, titrated to effect, and most animals are intubated after induction to provide oxygen and gaseous anaesthetic agents for maintenance.

Alfaxalone/alphadolone preparations have been used to anaesthetise several species of reptiles, and were most reliable in lizards and chelonia (Lawrence and Jackson, 1983). The author has also used alfaxalone (Alfaxan®, Vétoquinol, Buckingham, UK) as the sole agent to induce anaesthesia in lizards and chelonia.

Neuromuscular blocking agents provide immobilisation, but no analgesia, and IPPV will be required if they are used (Redrobe, 2004).

Anaesthetic maintenance

After induction, even very small reptiles can be intubated. Maintenance is usually achieved with volatile agents. PPV is often required as respiratory depression is common.

Recovery

If gaseous anaesthetics are used, these are switched off. Some injectable anaesthetics are reversible, for example medetomidine with atipamezole. The reptile patient is kept intubated until making spontaneous respiratory movements.

The high levels of oxygen supplied during anaesthesia will suppress the reptile's respiratory drive and continued oxygen provision during recovery will increase the time until spontaneous respiration (Diethelm, 2001). Two methods are used to reduce this, the first being to use carbon dioxide along with oxygen as carrier gases during anaesthesia. The second is to provide PPV with room air during recovery, for example using a respiratory or Ambu bag (see Chapter 1), rather than using 100% oxygen as is common practice in mammals.

Supplemental oxygen should be administered if the reptile patient has respiratory disease and hypoxaemia is suspected. Once the animal has been extubated, a facemask (with oxygen flow rate of 2–5 L/min) or nasal catheter (with oxygen at 0.5–3 L/min) can be used to provide oxygen if necessary. In cases with obstructive upper airway disease, a transcutaneous tracheal catheter can be placed to provide oxygen into the trachea. Oxygen should be humidified (see Fig. 1.11) to prevent drying of the respiratory passages (Schumacher and Yelen, 2006).

As the normal glottal position is closed in reptiles, the endotracheal tube is left in place until the animal is taking regular deep breaths, and oral and pharyngeal reflexes are present. In chelonia the forelimbs can be pulled out and pushed into the shell to cause respiratory movements. In all species, the use of small resuscitators (see Fig. 1.12) or Ambu®-bags (Ambu, Glen Burnie, MD) will enable ventilation with room air if required.

> Under no circumstances should the clinician blow directly into an endotracheal tube in a patient, as reptiles harbour many potentially zoonotic pathogens.

Heart rate and respiratory rate and pattern should continue to be monitored during recovery. Reflexes assessed during anaesthesia can also be monitored, including palpebral, corneal, foot and tail withdrawal. These parameters will allow the clinician to assess the progress of recovery from anaesthesia. If metabolic derangements or dehydration were identified pre-anaesthesia, it may be useful to repeat blood analysis to monitor these also during the recovery period.

The reptile should be kept within its POTR during recovery. If PPV is being performed outwith the vivarium, the use of heat pads and a hair-dryer (Fig. 12.9) will help maintain sufficient environmental temperature. Although in general warming reptiles increases their metabolism, there is no benefit to be gained from over-warming the patient, as the metabolic rate will increase along with oxygen requirements. During recovery the patient's depressed respiratory function may not be able to meet these requirements, resulting in hypoxaemia. If recovery is more prolonged than expected, doxapram can be administered as a respiratory stimulant.



Figure 12.9 • During recovery heat pads and hair-dryers may be used to warm reptile patients, such as this Veiled chameleon (*Chamaeleo calyptratus*). Care should be taken not to exceed the species' preferred optimum temperature range (POTR).

Recovery from anaesthesia is often prolonged in reptiles, due to their slow metabolism of drugs. If possible, reptile anaesthetics should be performed early in the working day, in order to ensure sufficient staff are available to monitor the patient during recovery. If the sole use of injectable agents such as ketamine is envisaged, the client should be forewarned that the pet is likely to require overnight hospitalisation post anaesthesia. This provides further opportunity for supportive care, such as fluids and analgesia, to be administered to the animal as necessary.

Provision of analgesia is vital for recovery. In the short term animals suffering discomfort or pain will be less likely to eat and, in the longer term, chronic pain will increase stress and, thus, affect other metabolic processes and the immune system.

To prevent drowning, aquatic species should not be allowed access to water until sufficiently recovered from anaesthesia (see Fig. 15.3). Similarly, group animals should not be returned to their companions until fully recovered, as attacks may occur.

Suggested anaesthetic protocols

In small animals or those that are severely debilitated, induction with an inhalational agent in a chamber (see Fig. 13.11) may be possible without pre-medication. Respiratory rate and pattern should be closely monitored to assess for

breath holding. If this technique is not possible, doses for injectable agents should be carefully calculated based on an accurate weight.

In larger animals injectable agents can be used either to cause sedation allowing mask induction or to induce anaesthesia, prior to maintenance with inhalational agents. The use of combinations of anaesthetic agents reduces the doses of other agents required, thereby reducing side effects. Propofol is the agent of choice to induce anaesthesia if intravenous access, for example a coccygeal or jugular vein, or intraosseous access is possible. If this is not an option, ketamine may be used alone or in combination with other agents intramuscularly to provide sedation. Butorphanol or buprenorphine is commonly used to sedate reptiles before induction with inhalational agents via a closely fitting facemask (Schumacher and Yelen, 2006).

Doses of anaesthetic agents in reptiles are often reported in large ranges. There are two reasons for this, the first being the wide difference in response to these drugs between species and individuals. The second reason is the great variation in condition of individual animals presented to the clinician for anaesthesia. It is for this latter reason that pre-anaesthetic assessment and stabilisation are so important in these animals. If in any doubt as to an animal's health status, use a combination of drugs at low doses or use drugs where doses can be altered or given to effect, for example inhalational agents or intravenous propofol.

- There is great variation in inter- and intra-species response to anaesthetic agents administered to reptiles.
- The clinician should ascertain the individual animal's health status and adjust anaesthetic doses accordingly.

Intramuscular injectable agents, such as ketamine, will be required in some species, including some chelonia or venomous snakes, to facilitate handling and enable induction of anaesthesia using other agents.

ANAESTHESIA MONITORING

Observations on the patient

Cardio-respiratory systems

As evidenced by the difficulties in initial clinical assessment of reptiles, monitoring the patient during anaesthesia is also problematic. A minimum of respiratory rate and pattern, and heart rate should be recorded during anaesthesia. There are great variations in species, and baseline data collected during the pre-anaesthetic period allow comparisons to be made for individual animals.

Anaesthetic agents produce cardio-respiratory depression, and often the respiratory drive is obliterated by a combination of drugs and oxygen administered during anaesthesia. Respiratory rate may, therefore, equal that produced by PPV. Anaesthetic records should differentiate between forced (PPV) and spontaneous ventilation.

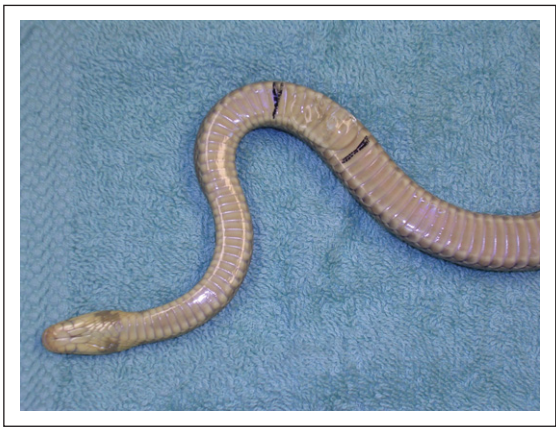


Figure 12.10 • Use tape or marker pen to mark the position of the snake's heart for ease of monitoring (shown here in a black rat snake [*Elaphe obsoleta obsoleta*]).

The heart rate and rhythm may be monitored visually or palpated in some patients but in most instances the use of an 8 MHz Doppler flow device (see Figs 12.6 and 13.12) assists greatly, particularly if surgical drapes cover the patient. The heart position varies between species. It is located in the cranial third of the body in snakes, but is quite mobile and it is helpful to mark the location with tape for repeated monitoring (Fig. 12.10). The chelonian heart is near the thoracic inlet (see Fig. 12.6), but a pencil probe is required to fit between the carapace and plastron in small species. In lizards, the heart is again usually near the thoracic inlet (see Fig. 13.2), but may be more caudally situated within the thoracic region in monitors. The carotid artery may be monitored in chelonia and lizards (see Fig. 13.12), or the coccygeal artery in lizards and snakes (Schumacher and Yelen, 2006). Small probes can be attached using adhesive tape to the skin for continuous monitoring, or probes with a larger footprint applied intermittently. Oesophageal stethoscopes may be of use in larger animals.

Nervous system

As with other species, various reflexes can be monitored to assess anaesthetic depth. Deepening of anaesthesia will reduce muscular tone. During induction, loss of the righting reflex is used to indicate anaesthesia. The palpebral reflex is also lost at this stage. The corneal reflex should be maintained during surgical anaesthesia; this reflex is lost at a deep plane of anaesthesia. The corneal and palpebral reflexes cannot be assessed in species with spectacles, including snakes and some lizards. Tail, toe and cloacal reflexes should also be monitored during anaesthesia (Schumacher and Yelen, 2006).

Snakes appear to lose muscular tone gradually, starting with the head and moving caudally. During recovery from anaesthesia, the reverse occurs, with tail tone returning first. Assessment of tail tone is, therefore, a useful measure of depth of anaesthesia in snakes.

Anaesthetic monitoring equipment

An 8 MHz *Doppler* flow probe is ideal for monitoring heart rate in reptiles. In smaller animals, it is well lubricated and pressed against the body wall in the position of the heart. For larger lizards or chelonia, a signal can be obtained from the carotid artery. *Oesophageal stethoscopes* are of use only in larger patients.

Electrocardiogram (ECG) can be used in reptiles, with leads positioned as in other species (Fig. 12.11). In snakes, electrodes are placed two heart lengths cranial and caudal to the heart (Girling and Hynes, 2002). To increase electrical conduction, needle probes can be placed subcutaneously. Alternatively, alligator clips are attached to skin and ECG gel applied (Schoemaker and Zandvliet, 2005). ECGs will only record electrical activity, and not mechanical activity; nonetheless, they can be useful in patients with cardiac disease (Schumacher and Yelen, 2006). Unfortunately, reference values do not exist for most species.

BOX 12.10 Stages of anaesthesia in reptiles (Brogard, 1987)

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Please refer to the printed publication.



Figure 12.11 • Electrocardiogram probes can be attached to needles inserted into limb muscles to improve contact, shown in this bearded dragon (*Pogona vitticeps*).

Direct arterial *blood pressure* can be measured, but is technically difficult and not routinely performed. A cut-down technique is used to access femoral, carotid or coccygeal arteries in lizards, chelonia and crocodilia (Schumacher and Yelen, 2006; Wellehan et al., 2004).

As discussed in the physiology section, reptile haemoglobin differs from that in mammals. For this reason, *pulse oximeters* designed to measure relative arterial oxygen saturation (SpO_2) will be calibrated for mammals, and absolute values will not relate to the reptile's SpO_2. Having said that, trends in oxygen saturation can be followed using these devices. In most reptiles, an oesophageal probe level with the carotid artery or a rectal probe can be used (Schumacher and Yelen, 2006).

Although arterial blood gas analysis is possible in reptiles, peripheral artery catheterisation is technically difficult and not routinely performed. Other *blood parameters* assessed prior to anaesthesia can be monitored during anaesthesia and in the recovery period. The maximum blood volume that can be withdrawn from a reptile patient is 0.5–0.8% of body weight (i.e. 0.5–0.8 ml per 100 g) (Heard et al., 2004), and this total volume should not be exceeded in any 2-week period.

End-tidal carbon dioxide concentration measurements are extremely useful for monitoring mammalian respiratory performance, but they are less useful in reptiles. The small size of most reptile patients necessitates the use of *capnograph* machines that sample 50 ml per minute. Reptile cardiac shunts will affect the measurement, and one study reports a lack of correlation between end-tidal and arterial carbon dioxide values (Hernandez-Divers et al., 2004). Nevertheless, trends in measured end-tidal carbon dioxide measurements may be useful during anaesthesia. A decrease may suggest airway leaks or obstruction, disconnection of patient from breathing circuit, or ventilator malfunction (Schumacher and Yelen, 2006).

PERI-ANAESTHETIC SUPPORTIVE CARE

Most pet species are used to being handled, but the clinician should be aware of handling techniques that will be safe for both the animal and handler(s). These techniques will vary between species, and special care should be taken when restraining ill or injured animals. (Other texts should be consulted for information on animal handling.)

Fasting

Pre-anaesthetic fasting is not necessary for most reptilian pets. The main exceptions are carnivores that may regurgitate food, or the recently fed snake that may have cardiorespiratory compromise due to the space-occupying mass within the gastrointestinal tract. Although many patients are receiving supplemental care, the clinician should avoid administering oral fluids shortly before anaesthesia, as aspiration may occur if they are regurgitated.

Fasting may alter gastrointestinal flora, and herbivorous species are not usually fasted before anaesthesia.

Non-herbivorous animals may be fasted from 18 to 96 h depending on the species (Redrobe, 2004).

Heating

During anaesthesia, the reptile patient should be maintained within its species-specific POTR. This is likely to involve heating of the room along with more localised heating of the animal. Heat pads, for example electric pads or those that can be warmed in microwave ovens, are useful. Circulating hot water blankets and forced air blankets are ideal, but gloves containing warm water are a cheaper alternative. Heat sources should usually be covered with a towel to prevent contact burns. Overhead lamps can also be used, but may prove uncomfortable for the clinician performing a procedure underneath.

Conversely, the patient should not be overheated. Temperatures over 42°C are fatal (Malley, 1997).

Fluids

Fluids should be given to all anaesthetised patients. In dehydrated or severely debilitated animals where hypotension is likely, the intravenous or intraosseous route should be used. Infusion pumps can be used for continuous administration of fluids, with syringe drivers being useful in smaller patients. Where this equipment is not available, an assistant can administer small boluses throughout the procedure. An extension set attached to an intravenous or intraosseous catheter allows intermittent fluid boluses to be administered easily during anaesthesia and the recovery period. Due to the small marrow space in reptiles, fluid boluses can only be of small volume. Placement of intraosseous catheters may cause damage to the patient, for example osteomyelitis if aseptic technique is not used for placement, or fractures in animals with bone pathology. Intraosseous catheters are discussed later in species subsections.

Maintenance fluid requirements are usually administered to reptiles at 5–10 ml/kg/h using a balanced electrolyte solution (Schumacher and Yelen, 2006).

Analgesia

Any animal that is suspected of having a painful condition or about to undergo surgery should be given analgesia. Opioid agents, such as butorphanol and buprenorphine, also have the advantage of providing mild-to-moderate sedation, and may reduce the doses of other anaesthetic agents required. Analgesia requirements should be assessed throughout anaesthesia; voluntary movement, or elevations in heart or respiratory rate during a procedure may indicate pain (Schumacher and Yelen, 2006). As with other species, pre-emptive analgesia should be provided where possible.

Multi-modal therapy is advisable for many procedures, to reduce possible side effects from one group of drugs. Commonly, a non-steroidal anti-inflammatory drug (NSAID) is used in conjunction with an opioid. Local anaesthetics may also be used topically or by local infiltration for certain cases, for example during orthopaedic or soft tissue surgery (see Fig. 13.5).

Cold narcosis is not recommended as a means of restraint for reptiles, as no analgesia is provided (Bennett, 1998).

Opioids and non-steroidal anti-inflammatory drugs

Often, pre-medication of an animal before surgery may involve the use of opioids such as butorphanol. Opioids are often used to manage acute pain in reptiles. NSAIDs are usually used in cases with chronic pain, for example gout. As in other species NSAIDs may exacerbate renal or gastrointestinal disease (Schumacher and Yelen, 2006). Gastrointestinal ulceration after NSAID administration has been reported anecdotally in chelonia.

Local anaesthetics

Local anaesthetics provide additional analgesia and also reduce anaesthetic agent requirements. Intercostal nerve blocks and interpleural anaesthesia have been reported in reptile coeliotomies. Local anaesthesia techniques in other species are applicable to reptiles. Lidocaine (lignocaine) and bupivacaine have been used in reptiles, often as a splash block, but they are not usually used as the sole agent for procedures. Lidocaine (lignocaine) has a more rapid onset, but bupivacaine is longer-acting (Schumacher and Yelen, 2006). As local anaesthetics may be toxic at higher doses, potentially causing arrhythmias and seizures, a maximum dose should be calculated (Table 12.5) to ensure that it is not accidently exceeded, particularly in small patients.

Table 12.5: Analgesic agents used in reptiles. Species variations exist in analgesic effect and duration of action (species-specific doses, where available, are listed in individual chapters)

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Bupivacaine	1–2[4]	Local	4–12	Beware toxicity; maximum total dose 4 mg/kg
Buprenorphine	0.02–0.20[3,4]	SC, IM	12–24	Analgesia and sedation

(*Continued*)

Table 12.5: (Continued)

DRUG	DOSE (mg/kg)	ROUTE	DURATION (hours)	COMMENT
Butorphanol	0.4–2.0[4]	SC, IM, IV	12–24	(pre-anaesthetic)
Carprofen	1–4[2,3]	PO, SC, IM, IV	24	Use half dose if repeat
Flunixin meglumine	0.5–2.0[2,4]	IM	12–24	–
Ketoprofen	2[2]	SC, IM	24	–
Lidocaine (lignocaine)	2–5[4]	Topical, local	–	Beware toxicity; maximum total dose 10 mg/kg
Meloxicam	0.1–0.2[2,4]	PO, IM, SC	24	Therapeutic levels may not be achieved with oral administration
Morphine	0.4–2.0[4]	SC, IM	12	Great species variability
Oxymorphone	0.1–0.2[4]	SC, IM	12–24	Great species variability, no effect in snakes[1]. Avoid if hepatic or renal dysfunction

Key: SC = subcutaneously, IM = intramuscularly, IV = intravenously, PO = orally

[1] (Bennett, 1991); [2] (Lawton, 1999); [3] (McArthur et al., 2004b); [4] (Schumacher and Yeten, 2006)

Table 12.6: Sedative and anaesthetic agents used in reptiles. Species variations exist in anaesthetic effect (species-specific doses, where available, are listed in individual chapters)

DRUG	DOSE (MG/KG)	ROUTE	SPECIES	COMMENT
Alkylphenol:				Dose to effect
• Propofol	5–10[1,23]	IV, IC	Snakes	Lower dose required if
	10[6,12]	IV, IO	Lizards	premedication administered
	12–15[10,26]	IV	Chelonia	
	10–15[19]	IV	Crocodilia	
Alpha-2-agonists:				Poor immobilisation if used alone.
• Medetomidine	0.1–0.15[5]	IM	Most	Reversible (with atipamezole,
• Xylazine	0.10–1.25[14]			yohimbine)
Alpha$_2$-antagonist:		IM, (IV)	–	Reversal of alpha-2-agonists
• Atipamezole	5 × medetomidine dose[13,25]			
Benzodiazepines:				Pre-anaesthetic. Minimal sedation
• Midazolam	2[2,4]	IM	Most	if used alone. Sedative in Red-eared slider, *Trachemys scripta elegans*, allowing minor manipulations
Dissociative agents:				Depth of sedation or
• Ketamine	5–90[9]	SC, IM, IV	Most	anaesthesia, and duration of recovery, are dose-dependent

DRUG	DOSE (mg/kg)	ROUTE	SPECIES	COMMENT
				Lower doses (10–20 mg/kg) used in most species as pre-medication
	55–88[3,4]			Surgical anaesthesia (induction 10–30 min, recovery 24–96 h)
• Tiletamine	See combination below	–	–	–
Inhalational agents:		Inhal	Most	Induction/maintenance
• Halothane	3–4%/1.5–2.0%[7,14]			Isoflurane preferred
• Isoflurane	3–5%/1–3%[8,16]			Advise intubate and IPPV
• Sevoflurane	Prn[15,22]			
Phenothiazine:			Most	Pre-anaesthetic, can be used with ketamine
• Acepromazine	0.05-0.50[17,21]	IM		
Steroids:				Good muscle relaxation
• Alfaxalone/	9[20]	IV	Snakes	Can repeat dose to extend anaesthesia
• Alphadolone	9–15[18,20]	IM, IV	Lizards, chelonia	Variable results in snakes
Combinations:				–
• Butorphanol + midazolam	0.4 + 2[5]	IM	–	Pre-anaesthetic
• Butorphanol + ketamine	0.5–1.5 + 10–30[24]		Chelonia	Anaesthesia, e.g. minor procedure
• Medetomidine + ketamine	0.1–0.3 + 10[11]		Most	Anaesthesia
• Tiletamine + zolazepam	4–10[4,5]		Most	Sedation; non-invasive procedures

Key: IC = intracardiac, IM = intramuscular, Inhal = inhalation, IO = intraosseous, IPPV = intermittent positive pressure ventilation, IV = intravenous, prn = dose to effect, SC = subcutaneous

[1] (Anderson et al., 1999); [2] (Bennett, 1991); [3] (Bennett, 1994); [4] (Bennett, 1996b); [5] (Bennett, 1998); [6] (Bennett et al., 1998); [7] (Boyer, 1992); [8] (Boyer, 1998); [9] (Carpenter, 2005); [10] (Divers, 1996); [11] (Divers, 1999a); [12] (Divers, 1999b); [13] (Fleming, 1996); [14] (Frye, 1994b); [15] (Heard, 1998); [16] (Jenkins, 1991a); [17] (Jenkins, 1991b); [18] (Lawton, 1999); [19] (Lloyd, 1999); [20] (Millichamp, 1988); [21] (Page, 1993); [22] (Rooney et al., 1999); [23] (Schaeffer, 1997); [24] (Schumacher, 1996); [25] (Smith et al., 1998); [26] (Stahl and Donoghue, 1997)

Table 12.7: Emergency drugs in reptiles

DRUG	DOSE (mg/kg)	ROUTE	INDICATION/COMMENT
Anticholinergics:			
Atropine	0.01–0.04[2,3,6]	SC, IM, IV, ICe	Bradycardia. Not effective at these doses in green iguana, *Iguana iguana*[4].
Glycopyrrolate	0.01[1,2]	SC, IM, IV	
Diazepam	2.5[5]	IM, IV	Seizures
Doxapram	5[1]	IM, IV	Respiratory stimulant; repeat every 10 min if required

Key: ICe = intracoelomic, IM = intramuscular, IV = intravenous, SC = subcutaneous

[1] (Bennett, 1998); [2] (Boyer, 1992); [3] (Frye, 1994b); [4] (Pace and Mader, 2002); [5] (Rossi, 1998); [6] (Schumacher, 1996)

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The most common post-anaesthetic problem with reptiles is apnoea. If respiratory arrest occurs, reptiles can survive long periods of hypoxia by converting to anaerobic metabolism. Maintenance with oxygen (or room air) ventilation once per minute may be sufficient to revive the patient after a prolonged period. As respiration is stimulated by low levels of oxygen, overventilation should be avoided, as this would depress respiration. Maintaining patients within their POTR is also important (O'Malley, 2005a).

A tracheostomy can be performed in an emergency, for example to bypass an upper airway obstruction (Schumacher and Yelen, 2006). Doxapram can be administered to stimulate respiratory movements in apnoeic animals.

The circulation should be supported with appropriate fluid administration, preferably intravenously in a critical patient.

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13 Lizard anaesthesia

INTRODUCTION

A diverse group of animals are found within the lizard suborder (Table 13.1), with many differences in external and internal anatomy, diet, and environmental requirements. These differences will almost certainly affect how a particular species is hospitalised and its supportive care, and may also affect the approach to anaesthesia.

Most lizards tolerate handling well. Larger species should be handled with care, particularly adult male iguanas during their breeding season from December to March (Barten, 2006). Smaller species, such as geckos, often have delicate skin that can easily be torn, and general anaesthesia using volatile agents may be required for physical examination.

Temperature

It is vital to know what species is being dealt with, as there is great variation in the preferred optimum temperature range (POTR) of lizard species. Although free-ranging animals may raise their body temperature on warmed rocks, radiant overhead (or wall) heating is preferable in captive animals, due to the risk of burns from direct contact with hot surfaces.

Cardiovascular system

The three-chambered heart lies at the pectoral girdle in most lizards (Figs 13.1 and 13.2). In monitors, it is more caudal (O'Malley, 2005).

Table 13.1: Families of lizards commonly kept as pets

FAMILY	EXAMPLES	COMMENTS
Iguanidae	Green iguana (*Iguana iguana*)	Whip-like tail; autotomy common
Agamidae	Bearded dragon (*Pogona vitticeps*) Frilled lizard (*Chlamydosaurus kingii*) Water dragon (*Physignathus cocincinus*)	–
Chameleonidae	Yemen veiled chameleon (*Chamaeleo calyptratus*)	Arboreal; drink from water droplets; take care to avoid damage to tongue when intubating
Gekkonidae/Eublepharidae	Leopard gecko (*Eublepharis macularius*)	Some gecko species have spectacles rather than eyelids; autotomy common
Varanidae	Bosc monitor (*Varanus exanthematicus*)	Heart quite caudal in body compared to other species
Scincidae	Blue-tongued skink (*Tiliqua* spp.)	–

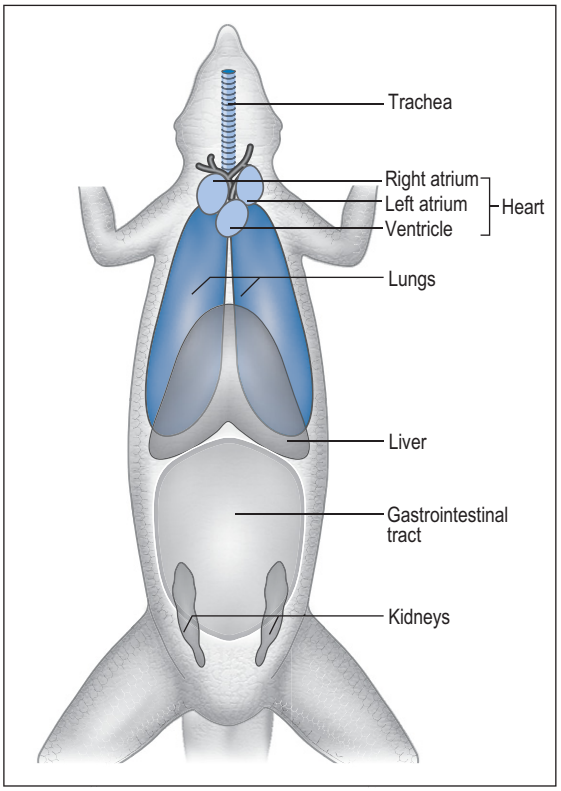


Figure 13.1 • Schematic showing lizard anatomy (ventral view).



Figure 13.2 • A Doppler flow monitor can be used to monitor the heart near the thoracic inlet in anaesthetised lizards, such as this green iguana (*Iguana iguana*). The heart is located more caudally in monitors. The endotracheal tube used in this patient is excessively long and will increase the respiratory dead space.

Pressure on the ocular globes in larger species causes a vagovagal reflex, with lowering of the heart rate and blood pressure. Non-painful procedures can be performed using this reflex (Bennett, 1972). The lizard will still respond to stimuli such as loud noise.

A renal portal system is present in lizards, receiving blood from the caudal vein and iliac veins. The efferent renal portal veins fuse to form the postcaval vein. There is a single ventral abdominal vein in lizards (Holz, 2006). As the ventral skin is often thin, the large ventral abdominal vein is accessible in most species. It is possible to inject medications into this vein (Table 13.2), but haemostasis may be difficult if haemorrhage occurs.

Respiratory system

In certain lizard species, for example the green iguana, nasal salt glands are used to excrete excess sodium and potassium salts. The resulting white powder at the nares is normal and not indicative of respiratory tract pathology (Barten, 2006).

The glottis is located at the base of the tongue (Fig. 13.3), but may be more rostral in some species, particularly carnivorous species, such as monitors. The normal resting position is closed, with the glottis opening only during inspiration and expiration. The tracheal rings in a lizard are incomplete. However, uncuffed endotracheal tubes are usually used, as most species are small and pressure necrosis may easily occur if a cuff was inflated excessively. The trachea bifurcates near the heart, into left and right bronchi (Murray, 2006).

Lizard lungs vary depending on the species (Fig. 13.4), with some having only a single chamber (unicameral), for example the green lizard (*Lacerta viridis*). Skinks have unicameral lungs with non-respiratory sacs caudally (similar to bird air sacs). Infection in these sacs can be difficult to eliminate, due to accumulation of exudates and poor vascularisation (Murray, 2006). Species such as the chameleon have paucicameral lungs, with more partitioning within the lungs and caudal dilatations (Fig. 13.5). In some species of chameleon, for example the Jackson's chameleon (*Chamaeleo jacksoni*), an accessory lung lobe is present in the ventral cervical region. In multicameral or multichambered lungs, faveoli are similar to mammalian alveoli (O'Malley, 2005). Iguanids have an anterior and posterior chamber in each lung (Schumacher and Yelen, 2006).

In paucicameral species and those with multicameral lungs, a postpulmonary septum divides the pleural from the peritoneal cavity. This membrane is not muscular and does not contain respiratory epithelium (Holz, 2006).

All vertebrae in lizards have ribs attached, except for those in the tail (Barten, 2006). The extent of these varies with species, with the chameleon coelomic cavity almost completely encased within the ribcage (Fig. 13.6). Contraction and expansion of the ribs cause respiratory movements. No diaphragm is present and outward movement of the intercostal, trunk and abdominal muscles causes inspiration due to negative pressure (and the converse for expiration). Lung walls also contain smooth muscle, which assists in inspiration in some species (Wood and Lenfant, 1976).

It is useful to observe a reptile's normal respiratory movements before anaesthesia, in order to assess depth of respiration and to mimic this movement during anaesthesia. Positive pressure ventilation (PPV) is useful in these animals, and their delicate lung structure makes overinflation and the possibility of rupture very easy.

Table 13.2: Routes of administration in lizards

ROUTE	SITE	NEEDLE/CATHETER SIZE	COMMENTS
Intracoelomic	Caudal right quadrant of coelom	22–25 ga needle	Rapid absorption; large volumes possible; risk of perforation of organs (less safe in conscious animals and reproductively active females)
Intramuscular	Anterior forelimb or hindlimb musculature	As small as possible for medication	Renal portal system may be important if hindlimb used Small volumes only
Intraosseous	Distal femur or proximal tibia	Intraosseous needle (with stylet), or hypodermic needle (with or without sterile surgical wire as stylet)	Ideal in collapsed patients where intravenous access not possible Possible to place catheter in distal femur without entering stifle joint in most species Equivalent to intravenous administration; can use to administer injectable anaesthetics or fluids
Intravenous	Ventral coccygeal vein	Long needle necessary in animals with thick tails	Vein larger near tail base, but avoid hemipenes in males Beware autotomy
	Jugular vein	Can catheterise in larger species with long necks 18–22 ga catheter in adult green iguana, *Iguana iguana*	Use local anaesthetic (e.g. 2% lidocaine (lignocaine)) before incise skin Maintain for maximum of 48–96 h Under anaesthesia
	Ventral abdominal vein	Small gauge needle. Can be catheterised	Haemostasis difficult if haemorrhage occurs
	Intracardiac		Emergency only
	Axillary venous plexus	–	Lymph dilution common
	Cephalic vein	Can catheterise in larger species	
Oral	Distal oesophagus or stomach	Use rubber or metal tube Pre-measure length to reach stomach	Ideal for administration of fluids and supplemental feeding; conscious animals only. (If the patient is alert, small volumes may be placed into the oral cavity to be swallowed)
Subcutaneous	Dorsally between scapulae, or lateral body	–	Small volumes only, particularly in species with high skin tension (e.g. chameleons) Avoid in species with fragile skin (e.g. geckos)

(Mitchell, 2006; Murray, 2000; Redrobe and MacDonald, 1999; Schumacher and Yelen, 2006)

Urinary system

Lizard kidneys are located dorso-caudally, lying within the coelomic cavity in most monitor lizards, but intrapelvic in most other species (Canny, 1998). In larger animals, clinical examination may include cloacal palpation to assess the intrapelvic region for renomegaly. Uric acid, urea or ammonia is produced to excrete nitrogenous waste; terrestrial species mainly produce uric acid to aid with water conservation.

Ureters from the kidneys empty into the urodeum portion of the cloaca. Most lizards have a urinary bladder, but in some species urine is stored in the distal colon prior to excretion (Davies et al., 1976). In those species with a

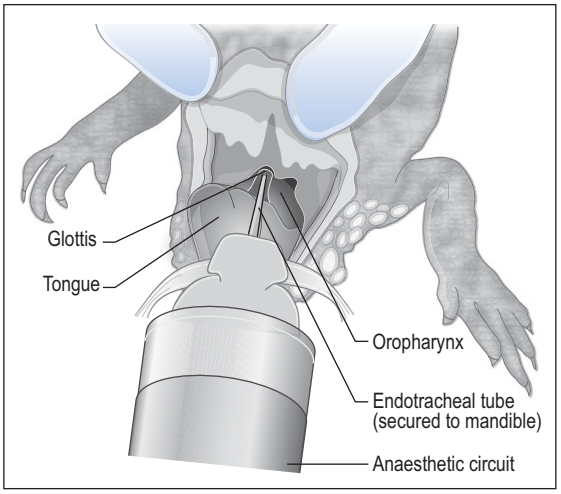


Figure 13.3 • Glottis is quite rostral in most lizards, such as this Chinese water dragon (*Physignathus cocincinus*), but may be more caudal in herbivore species.

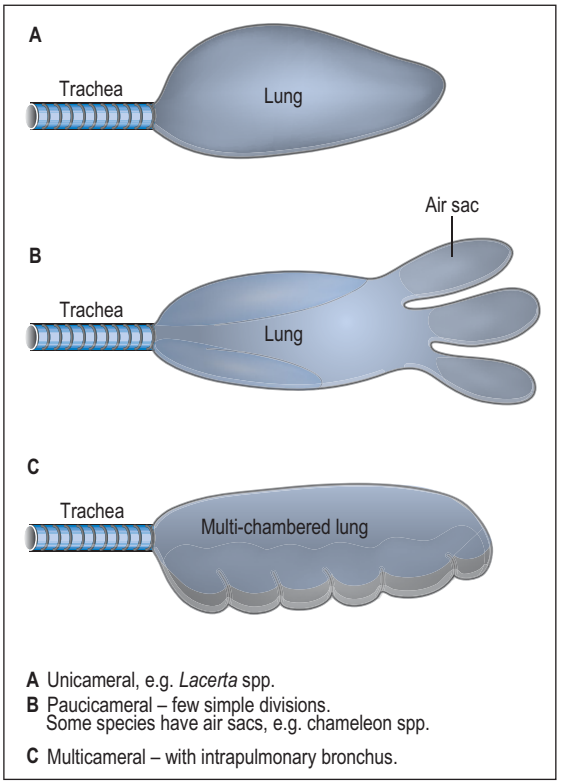


Figure 13.4 • Schematic showing lung types in lizards.



Figure 13.5 • Air sacs attached to the caudal lungs are exposed during coeliotomy to perform ovariectomy in this veiled chameleon (*Chamaeleo calyptratus*).

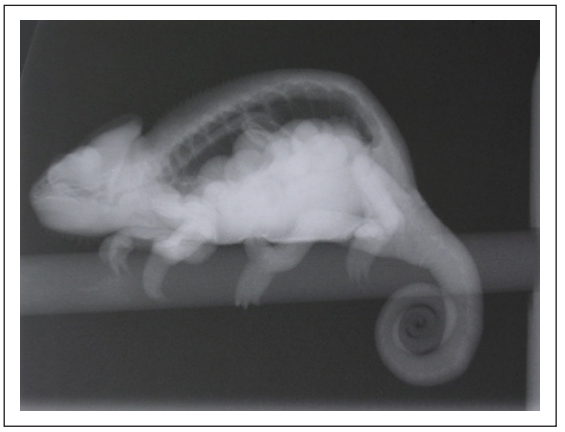


Figure 13.6 • Lateral radiograph of a veiled chameleon (*Chamaeleo calyptratus*) (with post-ovulatory egg stasis) showing the ribs enclosing the body. This is the same animal as in Figure 13.5 before surgery.

urinary bladder, the ureters do not connect with the bladder; rather the urethra connects the bladder to the urodeum (Beddard, 1904; Fox, 1977). Water can be reabsorbed from the bladder or colon. Animals fed diets containing excessive protein or deprived of water are predisposed to formation of cystic calculi (Barten, 2006). Nasal salt glands in herbivorous lizards aid in excretion of excess potassium salts, along with potassium urate salts in their urine (Dunson, 1976).

Branches from the aorta provide arterial blood to the kidneys. A renal portal system also directs blood from the tail and hind-limbs via the renal portal veins to the kidneys. Pelvic veins connecting to the iliac veins can divert blood around the kidneys (Holz, 2006, 1999).

Digestive system

The anatomy of lizard tongues varies between species, with most being relatively wide and fleshy, but some are highly specialised. The chameleon has a long sticky tongue,

monitors have a long forked tongue, while the gecko's is mobile enough to clean its corneas. Care should be taken not to damage the tongue during intubation, as it is a vital organ for prehension and swallowing of food.

In many species, including Agamidae and Chamaeleonidae, the teeth are acrodont and not replaced (Barten, 2006). Use a padded gag to avoid damaging dentition when opening the mouth for intubation or passage of a feeding tube. Protective gauntlets should be worn when restraining the venomous Helodermatidae, the Gila monster (*Heloderma suspectum*) and the Mexican beaded lizard (*Heloderma horridum*) and great care taken to avoid envenomation when examining the oral cavity.

Environmental temperature is important for digestive capacity, particularly in herbivorous reptiles. Certain species, including the green iguana and spiny-tailed lizard, require high temperatures for their microbial hindgut fermentations (Iverson, 1982). Gastrointestinal transit time is slow in herbivorous species, taking up to 140 h in lizards (O'Malley, 2005).

Diurnal lizards that do not eat vertebrate prey require ultraviolet (UV) (290–320 nm) light for vitamin D synthesis. They also benefit behaviourally and psychologically from UVA (320–400 nm) light (Frye, 1991; Gehrmann, 1994). UV light sources can be fluorescent tubes or bulbs, with some bulbs producing both UV light and heat. Lights producing 5% of their light as UV are recommended, as even these do not produce as much UV as direct sunlight. Most UV lights will continue to produce visible light when UV production has ceased, and, therefore, regular replacement is advised.

Herbivorous species are not usually fasted before anaesthesia. Smaller insectivorous or carnivorous species are fasted for 18 h before elective anaesthesia, and larger carnivorous species for 72–96 h (Redrobe, 2004).

Integumentary system

Autotomy is an escape mechanism practised by some species, whereby the lizard breaks off a portion of its tail mid-vertebra; it continues to move to distract a predator (including the unwary clinician). Care should be taken when handling species, including most Iguanidae, which may shed their tails in this manner, particularly when restraining the tail to access the ventral coccygeal vein. In this scenario, the clinician should grasp the pelvic girdle and hindlimbs along with the tail-base. If autotomy occurs the remaining tail should be cleaned, but not sutured, as regrowth (usually without calcification) will occur (Bellairs and Bryant, 1985).

TECHNIQUES

Routes of administration (Table 13.2)

Oral

The tube is pre-measured to mid-body. Use a mouth gag when gavage feeding lizards to prevent damage to the tube used, to the lizard's jaws (particularly if nutritional secondary hyperparathyroidism is present and pathological fractures likely), and to the operator. A wooden tongue depressor wrapped in bandage material (for example Vetrap®, 3M, St Paul, MN) can be used as a gag to allow safe gavage feeding. Alternatively a hole may be drilled in a wooden gag to facilitate passage of the tube. Visualise the glottis at the base of tongue, and direct the tube dorso-laterally into the pharynx and oesophagus.

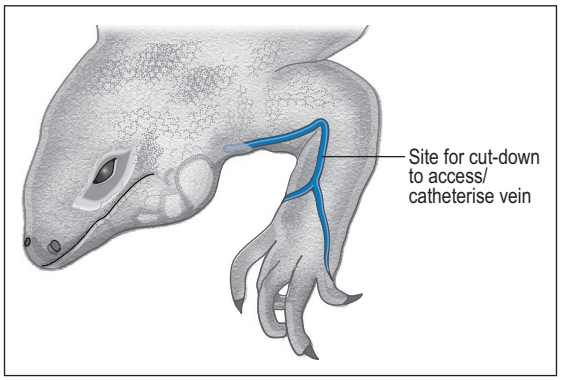


Figure 13.7 • Location of cephalic vein in lizard species.

Subcutaneous

The skin on the lateral flank is tented and the needle inserted at a shallow angle to avoid penetration into the coelomic cavity. Absorption is poor from this site. Some lizards (for example geckos) have particularly thin and friable skin, and the subcutaneous route should not be used in such species.

Intramuscular

The anterior muscles of the limbs are used (quadriceps and triceps). Pinch the muscle to stabilise it prior to injection.

Intracoelomic

Tilt the animal so one side is uppermost, allowing organs to drop away from the needle. Insert the needle at a shallow angle to the skin, paramedially to avoid the ventral abdominal vein. Aspirate before injecting, starting the procedure again (including fresh medication, syringe and needle) if air from a lung or air sac, or organ contents are aspirated.

Intravenous

Intravenous access is possible at several sites in lizards. Catheterisation requires use of a cut-down technique.

The *ventral coccygeal vein* is commonly used to administer drugs to induce anaesthesia. The needle is inserted midline a third of the distance down the tail at a steep angle (close to 90°) with the bevel facing up (see Fig. 13.10). When the needle contacts vertebral bone, withdraw slightly and aspirate to check positioning.

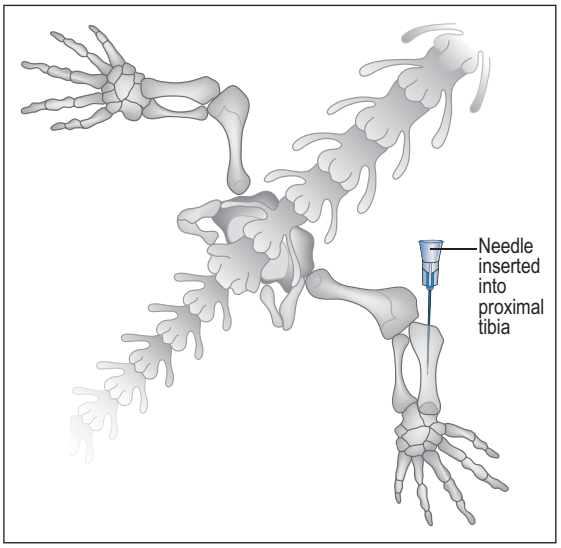


Figure 13.8 • Intraosseous catheter placement in the proximal tibia of lizard species.

A cut-down technique (and, therefore, anaesthesia) is required to reach the *jugular vein*. In the iguana, it lies on the ventro-lateral neck just caudo-ventral to the tympanic membrane in a line towards the shoulder. Some blunt dissection may be required to visualise the vein.

The *ventral abdominal vein* is notable for necessitating paramedical incisions for coeliotomy in the lizard. It lies relatively superficially in the midline of the ventral abdomen, and can be injected into or catheterised in larger animals. The *axillary venous plexus* lies caudal to the humerus near the shoulder joint.

Intracardiac injections may be required in an emergency. The heart is palpated and the needle inserted caudally into the pectoral inlet. Species such as monitor lizards have a more caudo-ventrally positioned heart, and the approach will be caudal to the forelimb.

The *cephalic vein* lies in a similar position to dogs and cats, coursing along the medial antebrachium (Fig. 13.7), and is accessible in larger animals. After surgical preparation of the skin, a small incision is carefully made in the skin overlying the vessel. A catheter can then be placed and attached to the skin with sutures or tissue glue before a light dressing is applied to protect the catheter from dislodgement (Mader and Rudloff, 2006). Fluids can be administered in boluses via a bung, or a continuous rate infusion using infusion pumps or syringe drivers. The ventral abdominal vein may also be catheterised, but maintenance is more difficult due to the location of the vein on the ventral body wall.

Avoid grasping lizard tails during restraint or venepuncture. Species that will undergo autotomy (tail loss) include the green iguana (*Iguana iguana*) and leopard gecko (*Eublepharis macularius*).

Intraosseous

Intraosseous catheters are easily placed in most lizards and provide access to the circulatory system equivalent to intravenous catheterisation. The most common site for placement is the proximal tibia, in the tibial crest (Fig. 13.8). As with intravenous catheterisation, a surgical preparation of the skin is performed. Analgesia should be administered, for example using local anaesthetic (taking care not to overdose and cause systemic toxicity) at the periosteum. Intraosseous catheters are available, but alternatives include spinal needles or hypodermic needles. The first two have the advantage of a central stylet, which prevents bone from blocking the needle during insertion. The latter is particularly useful in small species when the first two options are unavailable in small enough sizes, or where financial considerations preclude the use of the first two options. The needle should be a maximum of half the length of the bone.

To access the tibial crest, the stifle is flexed and the needle inserted in a distal fashion. With correct placement the bone will move in the direction of the needle when the hub is moved, there will be no resistance to injection of small amounts of fluid and there will be no soft tissue swelling due to injection into surrounding soft tissues. Radiographs can be used to confirm correct positioning.

The marrow space is small in reptiles and continuous rate infusions are preferable to bolus injections (Mader and Rudloff, 2006). Extreme care should be taken in patients where bone pathology is likely, for example nutritional secondary hyperparathyroidism, when pathological fractures may be induced during intraosseous catheter placement.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Injectable agents

The anaesthetic induction agent of choice in lizards is propofol. Most commonly, the drug is injected into the ventral coccygeal vein. In animals with a thick tail, a long needle is required. In species that undergo autotomy, the tail and body should be securely restrained to avoid this occurring. In patients with intraosseous access, this route can be used to administer the propofol.

Low doses of ketamine may be used to sedate patients before induction with propofol or volatile agents. Higher doses will induce anaesthesia, but have associated prolonged recovery times.

Tiletamine and zolazepam have been used intramuscularly to induce anaesthesia in green iguanas (*Iguana iguana)* (von Degerfeld, 2004). The author has also used alfaxalone intravenously in lizards.

Volatile agents

Mask or chamber induction with gaseous anaesthetic agents in lizards often results in breath holding, with a conversion to anaerobic respiration. Premedication with butorphanol (2 mg/kg intramuscularly) (Fig. 13.9) or buprenorphine of green iguanas causes sufficient sedation to allow mask induction (Schumacher and Yelen, 2006). The minimum



Figure 13.9 • Mask induction with isoflurane in a Chinese water dragon (*Physignathus cocincinus*) after pre-medication with intramuscular butorphanol.

alveolar concentration (MAC) of isoflurane in green iguanas is 2.1% (Mosley et al., 2003).

One study found more rapid induction and recovery from anaesthesia in green iguanas with sevoflurane compared with isoflurane, after butorphanol pre-medication in both groups (Hernandez-Divers et al., 2005). In the sevoflurane animals, muscle relaxation appeared greater and heart rates did not reduce as they did in the isoflurane group. Another study showed that butorphanol did not reduce the concentration of isoflurane required to anaesthetise green iguanas (*Iguana iguana)* (Mosley et al., 2003). The same author also demonstrated that the cardiac anaesthetic index of isoflurane was not affected by butorphanol in this species.

Anaesthetic maintenance

After induction, most lizards are intubated and maintained on volatile agents using intermittent positive pressure ventilation (IPPV) (either manually or mechanically).

Suggested anaesthetic protocols

As discussed in the introductory reptile chapter, many anaesthetic regimes have been used in lizards. As with other groups, the choice of protocol depends on species and health status.

In preference, lizards are induced using propofol intravenously (10–12 mg/kg) to allow intubation (Fig. 13.10).

In small patients or others where venous access is not possible, there are two options. The first is to use intramuscular anaesthetic agents to induce anaesthesia, often ketamine or ketamine combinations, but recoveries may be prolonged. A more balanced approach is to pre-medicate, for example with butorphanol (Hernandez-Divers et al., 2005) or low-dose ketamine, before induction using volatile agents using a facemask or chamber.

Healthier animals will often breath hold if subjected to volatile agents without any premedication, but severely debilitated animals may be induced using volatile agents alone (Fig. 13.11).

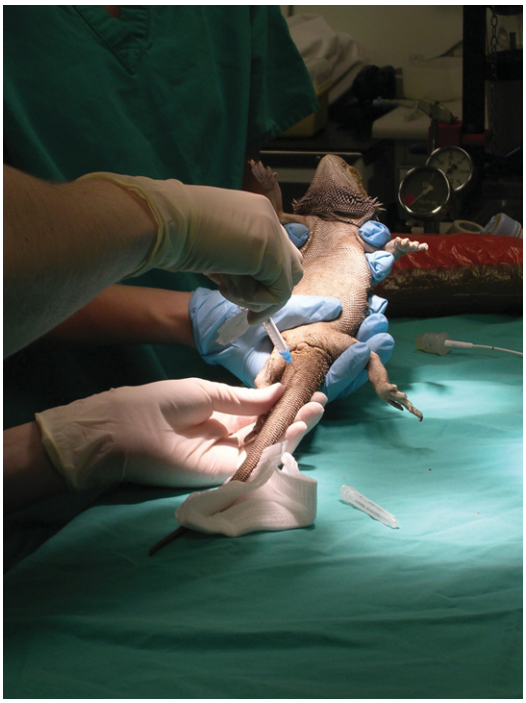


Figure 13.10 • Induction of anaesthesia in a bearded dragon (*Pogona vitticeps*) using intravenous propofol in the ventral coccygeal vein.



Figure 13.11 • Induction of a leopard gecko (*Eublepharis macularius*) in an induction chamber using isoflurane.

After induction, most patients can be intubated and anaesthesia maintained using volatile agents, such as isoflurane and sevoflurane. IPPV is usually required, as most anaesthetics depress respiration.

ANAESTHETIC MONITORING

If spontaneous respiration is present, the respiratory rate, depth and pattern should be observed. If anaesthesia

Table 13.3: Anaesthetic agents used in lizards (see also Table 12.6)

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Butorphanol	1–4[8]	IM	Premedication, reduces dose of induction agent; analgesia
Ketamine	5–10[7] 20–30[3]	IM	Premedication, reduces breath holding in chamber induction Sedation (lower dose than in other reptiles)
Ketamine + medetomidine	5–10 + 0.10–0.15[5]	(K) IM + (M) IM or IV	(Iguanas)
Isoflurane	4–5%[8] 1–3%[2]	Inhal	Induction of anaesthesia Maintenance of anaesthesia
Propofol	3–5[4,5] 0.3–0.5 mg/kg/min[8] 0.5–1.0[8]	IV, IO	Induction of anaesthesia, titrate to effect, apnoea common Higher dose required if not premedicated (<10 mg/kg[1]) Continuous rate infusion to maintain anaesthesia Intermittent boluses to top up anaesthesia
Sevoflurane	7–8%[8] prn[8]	Inhal	Induction of anaesthesia Maintenance of anaesthesia
Tiletamine/ zolazepam	4–6[8] 10–30[6]	IM	Sedation Anaesthesia (induction 8–20 min, recovery 2–10 h)

Key: IM = intramuscular, Inhal = inhalation, IO = intraosseous, IV = intravenous, prn = dose as required.
[1] (Bennett et al., 1998); [2] (Boyer, 1998); [3] (Faulkner and Archambault, 1993); [4] (Heard, 1998); [5] (Heard, 1999); [6] (Millichamp, 1988); [7] (Schumacher, 1996); [8] (Schumacher and Yelen, 2006)



Figure 13.12 • Doppler flow monitor on the carotid artery to monitor heart rate in a bearded dragon (*Pogona vitticeps*).

depresses respiration significantly, IPPV will be required. Heart rate and rhythm are best assessed using a Doppler probe over the heart (usually at the thoracic inlet [see Fig. 13.2], or more caudally in monitor lizards) or carotid artery (in larger patients [Fig. 13.12]).

Reflexes are monitored as for other species, with jaw tone and toe pinch withdrawal the most reliable for assessing surgical anaesthesia. The righting reflex is lost at a lighter plane of anaesthesia. The palpebral reflex can also be monitored (except in gecko species which have spectacles).

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Snake anaesthesia

14

INTRODUCTION

Although snakes evolved from lizards, there are many differences in their anatomy and physiology. There are 18 families of snakes. A great variety of snake sizes present the clinician with a variety of challenges, from obtaining venous access in the very small specimen to restraining the larger animal. This section will discuss general serpentine anatomy before approaching anaesthesia.

ANATOMY AND PHYSIOLOGY

General anatomy

In order to assess a snake's body condition, the normal appearance of that species should be known. The cross-sectional shape varies, but the dorsal spinous processes should be palpable. In obese animals these may not be palpable and in emaciated animals they will be overly prominent. Another assessment of condition is to test how much of the snake's body weight it can support unaided.

Most snake species (except Boidae) have a residual left lung, with only the right being functional. The trachea and heart lie in the cranial third of the snake's internal anatomy, with the lung(s) extending through both the cranial and middle thirds (Fig. 14.1).

The oesophagus passes through the cranial third, with the stomach found in the mid-section. For gavage feeding, the tube should be pre-measured to approximately half the length of the body. The ribs are numerous and continue for the length of the snake from head to cloaca. Intracoelomic injections, therefore, should be given along the ventro-lateral margin (see Fig 14.6).

Temperature

Preferred optimum temperature range (POTR) will vary depending on species – with most this is between 18°C

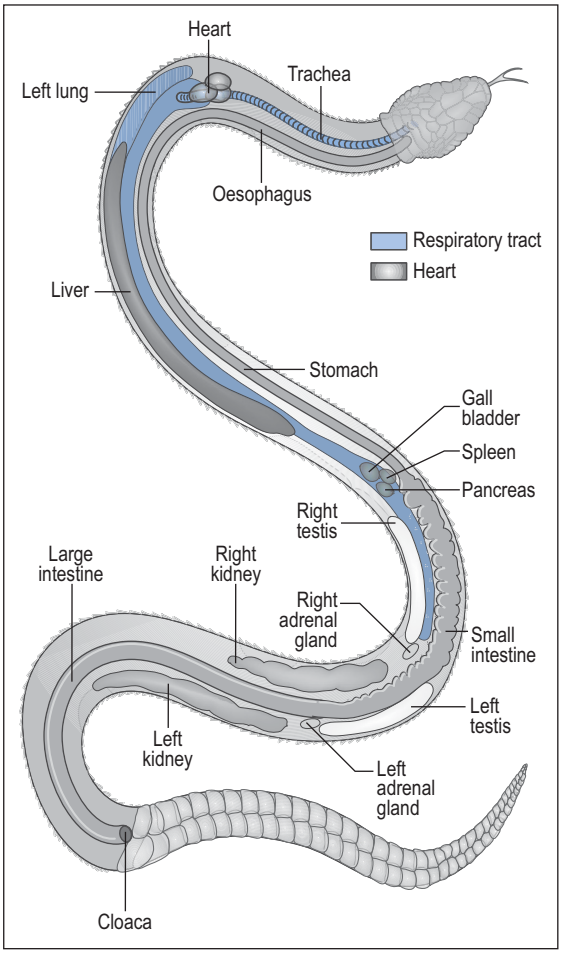


Figure 14.1 • Schematic showing organ location in snakes.



Figure 14.2 • Heart is stabilised to prevent displacement during cardiac puncture.

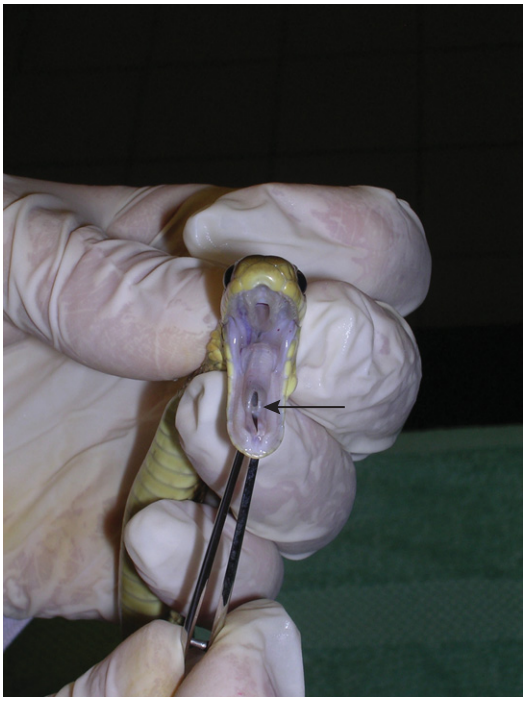


Figure 14.3 • Glottis is quite rostral in snakes, shown in this black rat snake (*Elaphe obsoleta obsoletea*).

and 34°C. Above 35°C heat stress occurs, with death at 38–44°C. Torpor occurs below 10°C, with death below 4°C (O'Malley, 2005).

All heat sources should be protected to prevent contact with the snake, as they may burn an animal that rests in contact with the heat for too long. Heat bulbs should, therefore, be covered with a grill (see Fig. 12.2). Heat mats should never be placed within an enclosure, under substrate, as the animal may dig down to lie directly on the mat.

Cardiovascular system

Snake hearts are similar to other three-chambered hearts in reptiles. Right-to-left and left-to-right shunting is possible, allowing mixing of oxygenated and deoxygenated blood (Hicks, 1998; Pough et al., 2004). The heart lies craniovental to the tracheal bifurcation. Owing to the absence of a diaphragm, the heart is fairly mobile and requires stabilisation with pressure both cranially and caudally before intracardiac injections (Fig. 14.2).

Oxygen requirements vary greatly in snakes. They increase postprandially to allow for increased metabolic requirements during digestion (Anderson et al., 2005).

Both renal and hepatic portal circulations are present in snakes, and as for other reptiles it is currently advisable to avoid injection of nephrotoxic medications into the caudal part of the body (Holz, 1999). There are no iliac veins in snakes, but an abdominal vein is present (Holz, 2006).

Respiratory system

The glottis is quite rostral in snakes (Fig. 14.3), allowing breathing to continue while they swallow prey and also relative ease of intubation even in conscious animals. Cartilaginous tracheal rings are incomplete, with a membranous region dorsally. In species such as gophersnakes (*Pituophis* spp.) the epiglottal cartilage is enlarged (Funk, 2006). Body positioning is important in clearance of exudates in the respiratory tract, as the mucociliary apparatus is inefficient (O'Malley, 2005).

In some snakes, i.e. the Acrochordidae, a portion of the trachea is open dorsally to a 'tracheal lung' with a respiratory surface (Stoakes, 1992). Most snake species have one functional lung, with the left being vestigial or absent (Table 14.1); the exception is Boidae that have two functional lungs. The right lung extends from the level of the heart almost to the right kidney (Funk, 2006). The cranial portion of the lung is single-chambered (unicameral) with faveoli lined by respiratory epithelium. The caudal third of the lung does not have a respiratory epithelium, and functions as an air sac (Perry, 1989).

The intercostal muscles control respiration. Several muscles control expiration. Inspiration is initially passive, following from relaxation of the expiratory muscles. The ribs are then elevated, causing a decrease in intrapulmonary pressure and active inspiration. When the inspiratory muscles relax and the lung(s) recoil(s), passive expiration occurs (Wood and Lenfant, 1976). As with other reptiles, the lung walls are thin and rupture is possible with over-zealous positive pressure ventilation (PPV).

Ophidian paramyxovirus (OPMV) has been associated with respiratory disease in certain snake species; serology may aid in diagnosis (Jacobson et al., 1981). Pneumonia may also be seen in Boidae with the retroviral inclusion body disease; diagnosis is based on histology of kidney or pancreatic biopsies (Schumacher et al., 1994).

Table 14.1: Families of snakes that may be kept as pets (only experienced herpetologists should keep venomous species, and staff safety at the veterinary practice is paramount if dealing with them)

FAMILY	EXAMPLES	COMMENTS
Boidae	Boas, e.g. boa constrictor (*Boa constrictor*) Pythons, e.g. Burmese python (*Python molurus*), royal python (*Python regius*)	Two lungs; two carotid arteries
Colubridae	e.g. corn snake (*Elaphe guttata*), common kingsnake (*Lampropeltis getula*), grass snake (*Natrix natrix*), milksnake (*Lampropeltis triangulum*)	One functional lung (right); single carotid artery (left) Most species harmless, but family includes rear-fanged venomous species
Elapidae	e.g. black mamba (*Dendroaspis polylepis*), king cobra (*Ophiophagus hannah*), taipan (*Oxyuranus scutellatus*)	One functional lung (right); single carotid artery (left) All venomous
Viperidae	e.g. Central American fer-de-lance (*Bothrops asper*), European adder (*Vipera berus*), Gaboon viper (*Bitis gabonicus*)	One functional lung (right); single carotid artery (left) Venomous

- The heart is located in the cranial third of the snake's body.
- The glottis is rostral in the mouth.
- Most snakes have a single functional lung, with a caudal air sac.

Urinary system

Serpentine kidneys are elongated and most are lobulated. The right kidney is more cranial in the body than the left, with both in the caudo-dorsal coelomic cavity. Snakes do not have a urinary bladder, so urine is stored in the distal colon or distal ureters (Canny, 1998; Holz, 2006).

The renal portal veins can be bypassed via the mesenteric vein, and thence to the liver (Holz, 2006).

Digestive system

Avoid trauma to the caudally facing teeth when examining a snake's oral cavity, for example during intubation or passing a feeding tube. Extreme caution should be taken in venomous species, even when anaesthetised, to avoid accidental self-envenomation.

The tongue in snakes lies in a sheath ventral to the glottis (Funk, 2006). The presence of a lingual fossa (Fig. 14.4) enables the snake to sample the air and environment with its tongue while the mouth is closed, and also allows the clinician to pass a feeding tube easily for administration of supportive fluids. When opening the mouth for examination, the clinician will note the unfused mandibular symphysis in snakes (Lock, 2006).

The oesophagus is relatively amuscular, and the cardiac sphincter is weak. After gavage feeding, the snake should be maintained in an upright position for at least 30 s to reduce the risk of regurgitation (O'Malley, 2005). Absorption of ingesta is slow, for example up to 5 days for a rat in a large snake.



Figure 14.4 • Rostral view of a juvenile Western hog-nosed snake (*Heterodon nasicus*) showing the lingual fossa. Feeding tubes can be passed through the fossa into the mouth, thus avoiding damage to the teeth.



Figure 14.5 • Subcutaneous injections are given between scales (shown in this black rat snake [*Elaphe obsoleta obsoletea*]). The skin is cleaned before needle insertion to reduce the risk of iatrogenic infection.



Figure 14.6 • Intracoelomic injection in a black rat snake (*Elaphe obsoleta obsoletea)*. The needle is inserted at a shallow angle to reduce the risk of visceral puncture.

All snakes are carnivorous. However, different species have different diets. Consuming larger prey requires a longer period of digestion and absorption, and larger snakes tend to feed less frequently (Funk, 2006). Snakes should be fasted before elective anaesthesia, to reduce the risk of cardio-respiratory compression from prey within the gastrointestinal tract. The period of starvation depends on the size of snake and usual frequency of feeding, but 72–96 h is usually advisable (Redrobe, 2004).

Integument

The ventral scales on snakes, the gastropeges, are larger and thicker than those dorsally and laterally. Snake scales are thickened regions of epidermis, with thin skin inbetween (O'Malley, 2005). Needles should be inserted into the thinner skin, to avoid damaging the protective scales (Fig. 14.5).

Special senses

The eyelids have fused in snakes to form a spectacle. It is not necessary to lubricate the eye during anaesthesia (as in other species), but care should be taken to avoid damage to the spectacle, as a lesion may lead to dysecdysis.

Olfaction is highly developed in snakes. If rodents or other prey species have recently been handled, wash hands or (preferably) wear gloves before handling a hungry snake that may taste prey on your hands and attack the scent. Similarly, snakes with infrared pits may rely on heat detection to strike at prey (O'Malley, 2005).

EQUIPMENT

Endotracheal tubes should be uncuffed. Those with a shoulder are useful in snakes to seal the glottis during anaesthesia. *Perspex tubes* may be used for restraint of venomous species during induction.

TECHNIQUES

Routes of administration

Oral

The feeding tube should be pre-measured to the level of the stomach, mid-body. Hold the snake with the head and neck vertical. Until the technician is experienced, it is advisable to open the mouth with a smooth plastic gag (for example, a small syringe) to visualise glottis. Once experienced, the technician can pass the tube into the closed oral cavity via the lingual notch, reducing the risk of dental trauma. Pass the lubricated tube into the caudo-lateral pharynx. Hold the snake with its proximal body elevated for 30 s to avoid regurgitation.

- Use a smooth mouth gag in snakes, for example the plastic or wooden shaft of a cotton-tip swab or a wooden tongue depressor.
- Bandage material will snag and damage the teeth.

Subcutaneous

Tent the skin before injecting between scales. Before any injection, the skin is surgically prepared to reduce introduction of pathogens. Needles are inserted between scales (see Fig 14.5).

Intracoelomic

The caudal coelomic cavity is accessed (Fig. 14.6). The needle is inserted at a shallow angle at the junction between the ventral scutes and lateral body scales. After aspiration to ensure organs have not been entered, fluids or medication can be slowly injected. If large fluid volumes are to be administered, they should be pre-warmed.

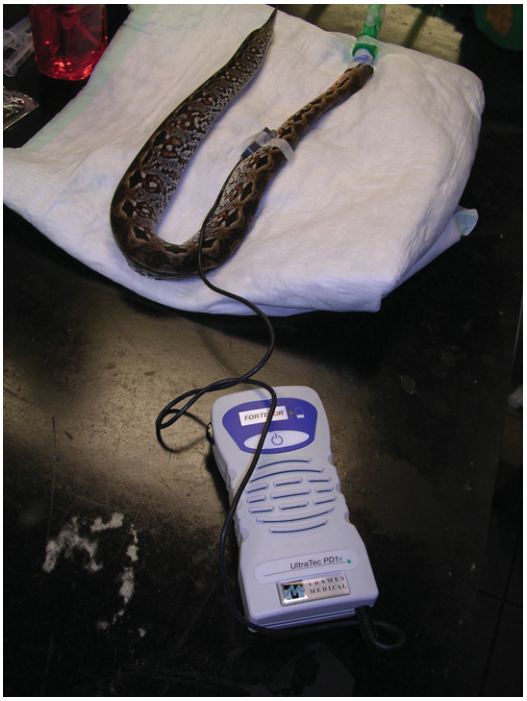


Figure 14.7 • A Doppler flow monitor can be used to assist location of the heart, which is located approximately one-third of the distance along the body length.

Intramuscular

The paravertebral muscles are used. Only small volumes can be administered via this route.

Venous access

Sites for venepuncture are scant in snakes due to their anatomical adaptations. Access is difficult in small animals or females with shorter tails.

Inserting the needle midline, directed caudally in males to avoid hemipenes, enters the *ventral coccygeal vein*. This vein is accessible in medium or large snakes. Sedation or anaesthesia may be required and haematoma is common (Murray, 2000).

Locate the *heart* by visualising the heartbeat or use a Doppler probe (Fig. 14.7). The heart is immobilised with gentle pressure cranially and caudally. After needle insertion, the syringe fills slowly with each heart contraction. There is a risk of haemorrhage associated with cardiac puncture, but the risk is small owing to the low blood pressure and slow heart rate in snakes (Murray, 2000). Extra care should be taken in animals weighing less than 300 g. Ideally, this route should be used for administration of drugs in emergencies only. In collapsed patients where venous access is not possible, an indwelling cardiac catheter may be placed.

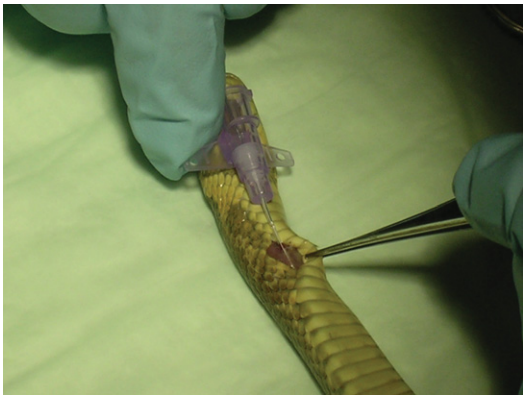


Figure 14.8 • A cut-down technique is required for jugular catheterisation in snakes such as this black rat snake (*Elaphe obsoleta obsoletea)*.

The *palatine vein* lies dorsally in the mouth, medial to the palatine teeth, and so is only accessible under general anaesthesia.

Intravenous catheters can be placed in snakes using a cut-down technique (Fig. 14.8). This is a useful technique in collapsed snakes, for administration of supportive fluids. Aseptic technique should be followed, with skin preparation before incision. The right jugular vein is most commonly used. The jugular vein is accessed approximately ten scales cranial to the heart. The vein traverses from a lateral position near the head towards the ventral heart. After surgical preparation of the skin, a small incision is made between the ventral scutes and lateral body scales. Some blunt dissection medial to the ribs is used to locate the vein under muscles. The catheter is inserted in a caudal fashion and secured with skin sutures.

In an emergency the heart can be catheterised, with the catheter inserted between scales into the ventricle. Haemorrhage and tamponade are possible, but unlikely due to the low blood pressure, slow heartbeat and thick ventricle in most snakes (Mader and Rudloff, 2006). A light dressing covering the catheter and bung prevents dislodgement during patient movements.

Intraosseous catheterisation is not possible in snakes.

Intubation

The glottis is rostral and relatively easily intubated in snakes (Fig. 14.9).

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

As with other reptiles, *propofol* is the anaesthetic agent of choice in snakes. However, venous access is difficult. The

Table 14.2: Routes of administration in snakes

ROUTE	SITE	COMMENTS
Intracoelomic	Caudal coelomic cavity	Technique similar to lizards
Intramuscular	Paravertebral muscles	Small volumes only
Intraosseous	N/A	Not possible in snakes
Intravenous	Ventral coccygeal vein	Difficult, particularly in species with short tail length caudal to cloaca
	Cardiac puncture	Catheterisation possible for debilitated patients
	Dorsal palatine vein	Under general anaesthesia
	Jugular vein (left or right)	Catheter placement possible
Oral	Mid or distal oesophagus	Manipulative procedures shortly after administration of oral fluids or medication are likely to induce regurgitation
Subcutaneous	Lateral body wall	Small volumes only; multiple sites may be required for larger volumes

(Mitchell, 2006; O'Malley, 2005; Redrobe and MacDonald, 1999)

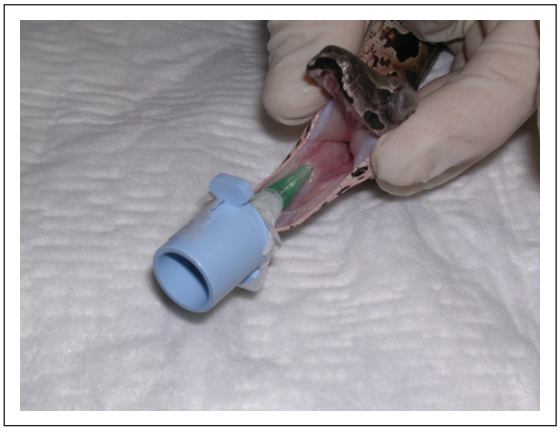


Figure 14.9 • The glottis is quite rostral in snakes (seen in this Madagascan ground boa [*Acrantophis madagascariensis*]), and easily intubated.

ventral coccygeal vein may be accessible in larger animals, but is short in females and is often small. If a jugular catheter has been placed, this can be used for administration of propofol. Cardiac puncture is not routinely used to administer anaesthetic agents, due to the risk of introduction of infection. The use of sedatives may facilitate intravenous access and administration of propofol.

In some specimens, it may be possible to *intubate* the conscious snake and induce anaesthesia with inhalational agents using PPV. Induction and recovery are rapid; however, this technique is stressful for the animal. Damage may be caused to the snake, either to the glottis during forced intubation, to the lungs and air sacs with over-zealous pressure, or to the subcutaneous tissues during restraint.

Although induction is possible using inhalational agents in a *chamber*, breath holding may occur and induction is not rapid. It is preferable to premedicate the snake to ease restraint and allow intubation. *Sedatives* used include butorphanol, low-dose ketamine or teletamine/zolazepam. Following intubation, anaesthesia is induced and maintained using inhalational anaesthetic agents.

If used as the sole anaesthetic agent, ketamine will cause respiratory depression, hypertension and tachycardia in snakes (Schumacher et al., 1997). Combinations can be used (see Table 12.6).

Anaesthetic maintenance

In general, snakes are intubated after induction and maintained using volatile agents. Repeated administration of injectable agents, such as ketamine, may excessively prolong recovery.

Suggested protocols

In preference, anaesthesia is induced with intravenous propofol. If intravenous access is not possible, the snake is pre-medicated with a low dose of ketamine to allow induction with volatile agents. This is done in an anaesthetic chamber, or by intubating the sedated patient and performing PPV with a volatile agent. Alternatively, a higher dose of ketamine may be used to induce anaesthesia, with or without other agents such as butorphanol.

Venomous snakes should be induced before intubation, to reduce the risk of self-envenomation. Protective equipment should be worn to reduce the risk to handlers. Induction can be performed either in a chamber with gaseous anaesthetic agents, or using injectable agents. Clear plastic tubes can be used to restrain the snake, with access for injections via small holes along the side.

Very small snakes pose difficulties to the clinician, as intravenous access and conscious intubation are not usually possible. Debilitated animals may allow induction in a chamber with inhalational anaesthetic agents. For more alert animals, injectable agents are used to sedate prior to induction with gaseous agents (Fig. 14.10) before intubation can be achieved. An accurate body weight is essential before administration of injectable drugs, diluting drugs if necessary and using insulin-type syringes with needles. Overdosage is a major concern, particularly as resuscitation techniques will also be limited in small animals. Intubation is performed after sedation, sometimes after administration of volatile agents in a chamber or via a mask.



Figure 14.10 • 60 g common garter snake (*Thamnophis sirtalis*) induced in a chamber with isoflurane after sedation with 10 mg/kg ketamine administered intramuscularly.

FORMULARY

Table 14.3: Anaesthetic and analgesic agents used in snakes (see also Table 12.6)

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Butorphanol	1–2[2]	IM	Analgesia; premedicant (improves muscle relaxation, e.g. with ketamine)
Diazepam	0.2–0.8[8]	IM	Muscle relaxation, used in conjunction with ketamine
Isoflurane	3–5%[9]	Inhal	Induction of anaesthesia
	<3%[9]		Maintenance of anaesthesia
Ketamine	5–20[9]	IM	Sedation; may require additional volatile agent to deepen anaesthesia in some patients before intubation is possible
	20–60[3,5]		Sedation (induction 30 min, recovery 2–48 h)
Propofol	5–10[1,7]	IV	Induction of anaesthesia, titrate to effect, apnoea common
	0.3–0.5 mg/kg/min[9]		Continuous rate infusion to maintain anaesthesia
	0.5–1.0[9]		Intermittent boluses to top up anaesthesia
Sevoflurane	8%[9]	Inhal	–
Tiletamine/zolazepam	3[6]	IM	Sedation
			Facilitates handling and intubation of large snakes
			Prolongs recovery
	10–40[4,6,10]		Induction of anaesthesia (in 8–20 min), recovery in 2–10 h, but variable
			Supplement with volatile agent for surgical anaesthesia

Key: IM = intramuscular, Inhal = inhalation, IV = intravenous

[1] (Anderson et al., 1999); [2] (Bennett et al., 1999); [3] (Boyer, 1992); [4] (Jenkins, 1991); [5] (Johnson, 1991); [6] (Millichamp, 1988); [7] (Schaeffer, 1997); [8] (Schumacher, 1996); [9] (Schumacher and Yelen, 2006); [10] (Schobert, 1987)

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Chelonian (tortoise, terrapin and turtle) anaesthesia

15

INTRODUCTION

Chelonia have been kept as pets for many years. Due to worldwide threats, many species are now protected, for example under Convention on the International Trade in Endangered Species (CITES) legislation. More pets now are captive-bred, but certain species are still traded internationally, and some aged specimens will have been traded prior to the establishment of CITES laws. Most species are difficult to keep in captivity, and unfortunately husbandry inadequacies often predispose disease.

In this text 'tortoise' refers to terrestrial species, 'terrapin' to freshwater semi-aquatic species, and 'turtle' to marine and aquatic species.

ANATOMY AND PHYSIOLOGY

Temperature

The table at the start of the reptile introductory chapter lists preferred optimum temperature ranges (POTR) for some species (see Table 12.1). For most terrestrial species this is between 26°C and 38°C, and for aquatic or semi-aquatic animals between 25°C and 35°C (Boyer and Boyer, 2006). Most tortoises bask to absorb heat through their carapace (dorsal shell), and so heating should be provided from above or to one side of their enclosure. Heating appliances include lamps and mats. In order to produce a temperature gradient (or range) across the vivarium, heaters should be predominantly at one end. This will provide a basking area, but supplemental background heating may also be necessary to maintain the lower end of the POTR. Ambient temperature for hospital enclosures should be 24–32°C for most species. For aquatic species, submersible water heaters may be required to maintain water temperature.

With all heaters, ensure that the chelonian cannot contact them, either by raising them above the substrate or using a barrier cage. Thermostats are useful to help maintain a constant temperature in the enclosure, but are not 100% reliable. Digital thermometers should be used to monitor both air and water temperatures in chelonian enclosures.

Cardiovascular system

The chelonian three-chambered heart lies cranio-ventrally in the coelomic cavity, near the pectoral inlet. As discussed above (see Chapter 12), the three-chambered heart still

Table 15.1: Families of chelonia that may be kept as pets

SUBORDER	FAMILIES	EXAMPLES	COMMENTS
Cryptodira (hidden-neck)	9	Leopard tortoise (*Geochelone pardalis*) Map turtles (*Graptemys* spp.) North African tortoise (*Testudo graeca*) Pig-nosed turtle (*Carettochelys insculpta*) Slider (*Trachemys scripta* subspp.)	Can retract head within shell
Pleurodira (side-neck)	2	African side-necked turtles (*Pelusios* spp.) Australian snake-necked turtles (*Chelodina* spp.)	Unable to retract head, but fold it sideways

maintains functionally separate pulmonary and systemic circulations (O'Malley, 2005).

The renal portal system is present in chelonians, but as in other reptiles its importance is not known. As with other species, nephrotoxic or renally excreted drugs are usually administered in the cranial half of the body (Boyer and Boyer, 2006).

The two chelonian abdominal veins are linked by a transverse anastomosis (Holz, 1999).

Respiratory system

Normal chelonian respiration is via the nares. The glottis is at the base of the fleshy tongue (Fig. 15.1). In cryptodiran species the trachea bifurcates relatively proximally, allowing respiration when the head is retracted within the shell. Short endotracheal tubes should be used to ensure administered gases enter both bronchi. In pleurodiran species, the trachea is longer due to the extended length of the neck. As the cartilaginous rings of the chelonian trachea are complete, non-cuffed endotracheal tubes should be used.

The lungs attach ventrally to the carapacial periosteum, from the pectoral to the pelvic limb girdles. The paired lungs are compartmentalised (multicameral), with a single primary bronchus each side (Perry, 1989). As in other reptiles, the respiratory surface area is small compared to mammals (Wood and Lenfant, 1976). Internal ridges and septa increase the surface area in the sac-like lungs, with faveoli opening into the central air space.

As the pectoral and pelvic girdle are within a chelonian's 'ribs', they cannot breath by moving these bones (McCutcheon, 1943). Respiratory movements are enabled by a combination of techniques. In most species, contraction of testocoracoideus and obliquus abdominis muscles enlarges the coelomic cavity leading to inspiration, and contraction of diaphragmaticus and transversus abdominis reduces the coelomic space and causes expiration. In *Testudo graeca* (North African tortoise), pectoral girdle movements are the predominant respiratory muscles (Wood and Lenfant, 1976). Outward movement of the fore- and hind-limbs tenses the septum horizontale, a non-muscular membrane which separates the airspace from the coelomic cavity. The negative pressure induced causes lung inflation. Movement of the limbs within the coelomic cavity relaxes this membrane, and also increases positive pressure on the gastrointestinal tract and thence the lungs, expelling air from the lungs (Gans and Hughes, 1967; Wood and Lenfant, 1976). Head and neck movement in and out of the shell also aids respiration (Boyer and Boyer, 2006).

Some aquatic species can respire across their skin, pharyngeal mucosa and/or cloacal bursae. Although aquatic species usually surface to breathe, many species can respire across their cloacal membrane sufficiently during periods of low activity, for example during underwater hibernation (brumation) (Cann, 1998; Davies, 1981).

An inefficient mucociliary clearance of foreign material in the airways and the caseous nature of inflammatory exudates in chelonia (Junge and Miller, 1992; Murray, 1996) mean that these species are susceptible to pneumonia. A cure is unlikely and pneumonia carries a poor long-term prognosis. However, their ability to respire anaerobically enables chelonia to survive severe respiratory disease (McArthur et al., 2004).

The dive reflex in chelonia is used physiologically when a low oxygen environment is encountered. The animals become apnoeic and convert to anaerobic respiration, as well as using cardiac shunting to divert blood from the pulmonary vessels (McArthur et al., 2004). Induction with volatile anaesthetic agents alone is usually not possible in these species.

Mycoplasma spp. have been associated with upper respiratory disease and pneumonia in tortoises (Jacobson, 2000; Mader, 1991). A herpesvirus has also been linked to upper and lower respiratory tract disease in chelonia (Junge and Miller, 1992; Origgi and Jacobson, 1999). *Mycoplasma* culture and viral isolation are difficult. Enzyme-linked immunosorbent assay (ELISA) testing has been developed for *Mycoplasma agassizii* in certain species. Polymerase chain reaction (PCR) assays can be used to diagnose *Mycoplasma* and herpervirus infections (McArthur, 2004c).

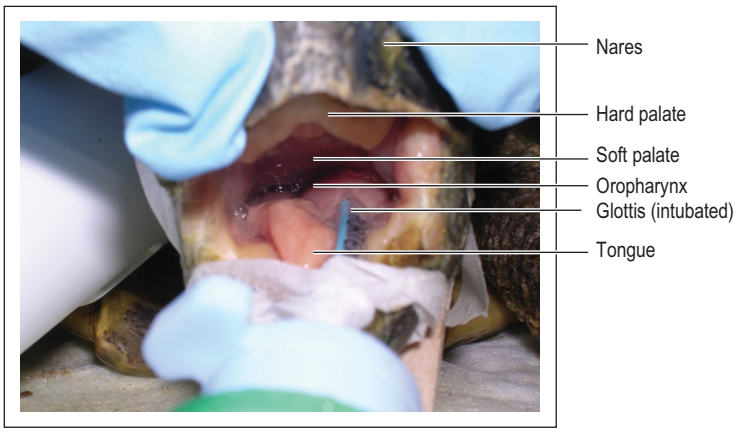


Figure 15.1 • Open-mouth view showing intubated *Testudo* sp., with the glottis at the base of the fleshy tongue.

Urinary system

The paired lobulated kidneys lie on the ventro-caudal carapace, caudal to the acetabulum in most species (Frye, 1991). Terrestrial species produce uric acid and urate salts to conserve water whilst excreting nitrogenous waste; aquatic species tend to excrete ammonia and urea (Davies, 1981).

The chelonian bladder may be a single large sac or may have bilateral accessory bladders (Holz, 2006). The ureters enter the bladder neck directly in chelonia (compared to other reptiles where they enter into the wall of the urodeum). A urethra then connects the bladder to the urodeum (Canny, 1998). The urinary bladder is used to store water in terrestrial species, with reabsorption of water (and excreted drug metabolites) possible from the cloaca, colon or urinary bladder (Davies, 1981). Cloacal bladders may also have respiratory epithelium in some species, necessary for periods of underwater hibernation (Fox, 1977).

Water should always be provided for hospitalised chelonia. Bowls should be deep enough to allow the animal to dip its head to drink. Tortoises benefit from a period of daily soaking in warm water or electrolyte solution to encourage drinking and defecation (see Fig. 12.8). Debilitated animals should be monitored to avoid drowning.

Gastrointestinal system

Chelonia are herbivorous or omnivorous (see Table 12.1). Terrestrial species are usually herbivorous, while aquatic species are usually carnivorous or omnivorous (Boyer and Boyer, 2006; Pritchard, 1979). Their gastrointestinal transit time varies between species, taking up to 4 weeks, and is significantly affected by factors such as temperature, dietary fibre and water content, and feeding frequency (King, 1996; Lawrence and Jackson, 1982).

Poor water quality will predispose various disease processes. It is preferable to feed aquatic species in a separate tank to their main enclosure to maintain water quality in the main aquarium. Many animals will void urine, urates and faeces around the time of feeding. Voided material should be removed from the water.

Non-herbivorous chelonia are usually fasted for 18 h before elective anaesthesia to reduce the risk of regurgitation and aspiration (Redrobe, 2004).

Integumentary system

The presence of a shell provides protection from predation for these animals. However, procedures performed routinely in other species are difficult or not possible, including limited clinical examination and restriction of venous access sites. A great variation in basic anatomy will affect which procedures may be performed, with due regard paid to staff safety when dealing with large carnivorous species, for example the snapping turtle (*Chelydra serpentine*).

The chelonian shell comprises a dorsal carapace and ventral plastron, although the exact shape and consistency will vary between species. For example, box turtles (*Terrapene* spp.) have hinges in their plastron, while hinged-back tortoises (*Kinixys* spp.) have a caudal carapacial hinge. These variations often restrict the clinician's access for examination and administration of medication. The anatomy of this group of reptiles, therefore, poses some challenges for treatment (Fig. 15.2).

Although newspaper will be used as the substrate in most hospital enclosures, most species of tortoise benefit from being able to hide or dig. For species from drier climes shredded newspaper allows some refuge, but for species requiring higher humidity a box or cat litter tray with damp soil and/or sand will allow more natural behaviour. A hide box should always be provided. Aquatic and semi-aquatic species should be provided with a dry area on which to haul out and bask (Fig. 15.3) (Boyer and Boyer, 2006).

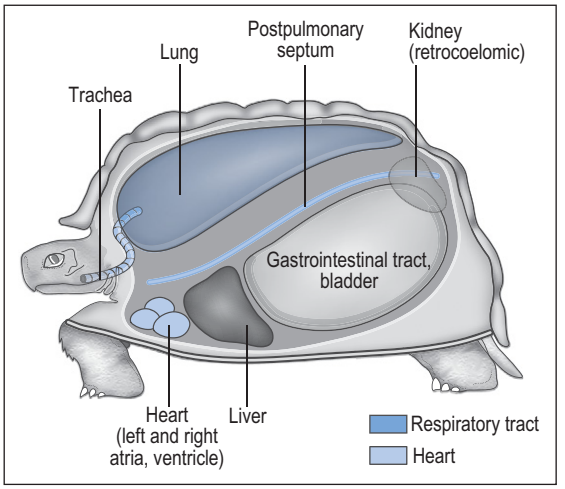


Figure 15.2 • Schematic lateral view of a chelonian showing the location of major organs within the shell.



Figure 15.3 • Aquatic and semi-aquatic species should be provided with a dry haul-out area during hospitalisation. The patient should not be returned to water until sufficiently recovered from anaesthesia. This pig-nosed turtle (*Carettochelys insculpta*) is not conscious enough to swim and may drown if left unattended.

Metabolism

As with other reptiles, the metabolic rate in chelonia is relatively low. The rate becomes much lower during periods of hibernation, in part due to the lower environmental temperatures during this time. This effect should be considered before administration of drugs. Animals recovering from illness should not be hibernated, as the immune function and healing will also be reduced.

TECHNIQUES

Routes of administration

Oral

Small volumes of medication can be syringed directly into the oral cavity or mixed with food if the patient is eating well. It is more usual to administer medication and fluids via gavage to ensure dosing success.

Pre-measure the rubber or metal tube to the level of the stomach, which lies mid-plastron. Restrain the patient's head in extension (straightening the oesophagus). Use a mouth gag – a finger in the mouth commissure is sufficient in most terrestrial tortoises, but a metal gag may be more appropriate in larger or stronger animals. Pass the tube dorso-laterally in the oro-pharynx to avoid the glottis (Fig. 15.4A). Observe for regurgitation during dosing. Large volumes of fluids should be pre-warmed to prevent body cooling.

This route should be used in conscious animals only. In less cooperative patients it is possible to pass the tube with the neck retracted, using a flexible tube and a mouth gag.

Subcutaneous

This route is rarely used in chelonia, as absorption is slow. The axillary or inguinal regions may be usually used, tenting the skin before injection.

Intramuscular

Muscle groups anterior to the humerus or femur are used. The limb is held and the needle inserted perpendicular to the skin (Fig. 15.4B). Only small volumes can be administered via this route.

Epicoelomic

Fluids can be administered into the potential space between the plastron and pectoral musculature (Fig. 15.4C) (McArthur, 2004b).

Intracoelomic

Access is via the prefemoral fossa, near the site where skin attaches to the plastron-carapace bridge. Tilt the animal so viscera fall away from the needle. Insert the needle into the uppermost side to enter the coelomic cavity (Fig. 15.4D). There is a risk of organ penetration and particular care should be taken in reproductively active females, as a sterile peritonitis may result from follicle or ova penetration. Large volumes of fluids should be pre-warmed before intracoelomic injection.

Intravenous

Several venous sites are present in chelonia. The choice will depend on the species and size of the animal, and the reason for access.

- Most terrestrial tortoises can be restrained with the head extended to access the jugular vein.
- It may be more difficult to grasp the head in turtles and terrapins; the subcarapacial vein is a useful alternative for venepuncture in these species.
- The subcarapacial vein is also accessible in very small chelonia.
- Species such as the African spurred tortoise (*Geochelone sulcata*) and box turtles (*Terrapene spp.*) often require sedation with intramuscular agents before venous access is possible.

The *jugular vein* is most commonly used for venepuncture in chelonia. The vein lies on the lateral neck, extending caudally from the tympanic membrane towards the thoracic inlet. The vein is quite superficial, with the right jugular vein often larger than the left. To access the vein the head is extended and the vein is raised by applying pressure at the base of neck laterally (Fig. 15.4E).

Although injections are often given through the skin into the jugular vein, it is easier to use a cut-down technique before catheterisation to expose the vein. Catheters of 20–26 gauge can be inserted, depending on body size. A cut-down technique may allow for easier catheterisation. The catheter is inserted approximately one-third the length of the neck caudal to the tympanic membrane, and should be approximately half the length of the neck. The catheter is attached as in lizards using skin sutures or tissue glue, and a bung used to allow fluid administration. If free-flowing giving sets are used, flow may cease when the chelonian's neck is retracted. Small syringe-drivers can be attached to the carapace in larger specimens to allow ease of movement (Mader and Rudloff, 2006).

The *subcarapacial vein* is ventral to the carapace in the midline. A needle is inserted at a 45° angle into the skin near the carapace. This vein cannot be catheterised, but is a useful site for venous access, particularly in small patients or in animals where the head cannot be easily extended (including many aquatic species).

The small *dorsal coccygeal vein* is accessible for phlebotomy or injection of small volumes of medication. Due to the location, it is important to clean the skin thoroughly before using this route. The tail is extended caudally, and the needle inserted midline at a steep angle until bone is reached. The needle is then withdrawn slightly and the syringe aspirated to check positioning. Inadvertent lymph vessel access is common at this site.

Access to the dorsal occipital venous plexus is possible from a lateral approach, but is rarely used (O'Malley, 2005).

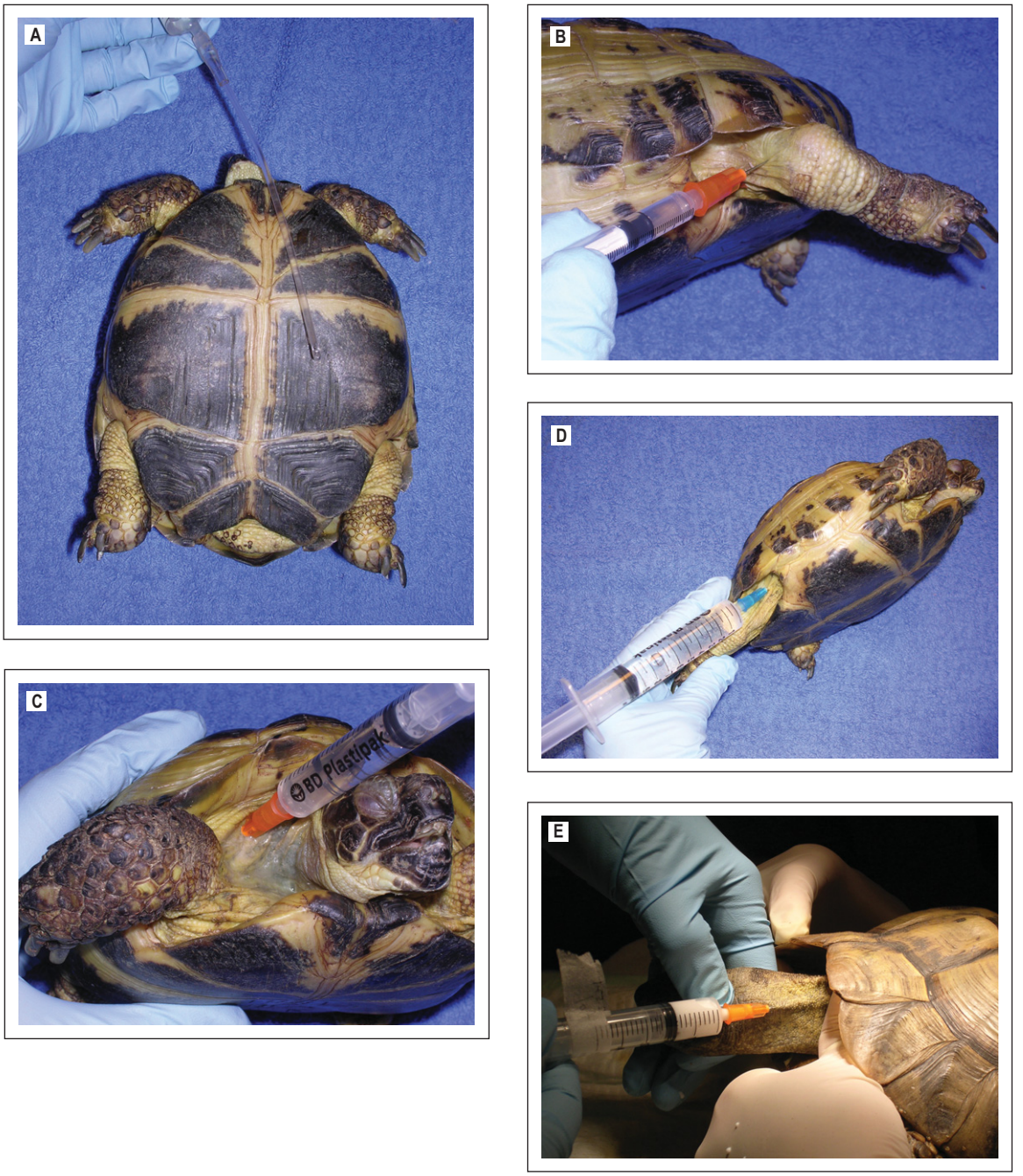


Figure 15.4 • Routes of administration in chelonian. (A) Premeasure the tube before gavage feeding. (B) Intramuscular injection in the quadriceps muscle. (C) Epicoelomic injection. (D) Intracoelomic injection. (E) Induction of anaesthesia by intravenous injection of propofol into the jugular vein of a Hermann's tortoise (*Testudo hermanni*).

Intraosseous

This is difficult in chelonian species. The distal femur or proximal tibia can be used in some animals. Femur access is difficult due to the sigmoid shape of this bone and intraosseous catheters in limbs may be dislodged by withdrawal of the limb within the shell. Access to the long limb bones is also restricted by the patient's propensity to withdraw the limb, but may be useful in debilitated patients. Additional problems are the thick cortices and small marrow cavities of chelonian bones (Mader and Rudloff, 2006).

Table 15.2: Routes of administration in chelonia (Fig. 15.4)

ROUTE	SITE	COMMENTS
Epicoelomic	Potential space between plastron and pectoral muscles	Absorption good Can administer 1–2% body weight in fluids, in divided doses, over 24 h
Intracoelomic	Prefemoral fossa	Rapid absorption Pre-warm large volumes of fluids
Intramuscular	Muscle groups anterior to humerus or femur	Can be difficult in species that withdraw limbs into closed shell, for example box turtle (*Terrapene* spp.)
Intraosseous	Cranial or caudal limits of bone bridge connecting carapace and plastron; distal femur or proximal tibia	Provide analgesia
Intravenous	Jugular vein Subcarapacial vein Dorsal coccygeal vein	Jugular vein may be catheterized
Oral	Encourage to drink by bathing Use rubber/polyvinyl chloride (PVC)/metal tube to reach stomach (mid-plastron level) Small volumes directly into mouth	Useful for administration of fluids and nutritional support
Subcutaneous	Axillary or inguinal region	Slow absorption

(McArthur, 2004b; Mitchell, 2006)

Another option is to access the bone within the shell, usually by inserting a needle into the cranial or caudal limits of the bony bridge between carapace and plastron. In larger animals an orthopaedic drill may be required to gain access to the medulla in this bone before insertion of a needle. As with other catheters, aseptic technique should be used for placement of the needle.

PRE-ANAESTHETICS

Pre-medications may be used solely for the purpose of sedation or as part of a balanced anaesthetic protocol in chelonian species (see Table 15.3).

Ketamine is often used to sedate chelonian species that are difficult to examine or to obtain venous access, for example in box turtles (*Terrapene* spp.) or African spurred tortoises (*Geochelone sulcata*). This drug has a relatively wide safety margin, but may be dangerous in dehydrated or debilitated tortoises. The effects are dose-dependent, with 20 mg/kg producing sedation in most species. *Benzodiazepines*, such as diazepam and midazolam, are often administered with ketamine to produce good muscle relaxation with sedation. Another dissociative–benzodiazepine combination is tiletamine–zolazepam – lower doses of this produce sedation.

The *phenothiazines* acepromazine and chlorpromazine have been used as pre-medicants in chelonia, administered intramuscularly. The addition of acepromazine to ketamine sedation reduces the ketamine dose required and allows for a more rapid induction and recovery than ketamine alone. Chlorpromazine is rarely used, but has anticholinergic as well as anxiolytic properties that may be beneficial.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Volatile agents

If anaesthesia is attempted in the conscious animal using inhalational anaesthetic agents alone, chelonia are very likely to breath hold, particularly aquatic species – undergoing the 'dive reflex' and switching to anaerobic respiration. Different species will be able to breath hold for varying time periods, with aquatic animals capable of maintaining anaerobic respiration for the longest time. An exception may be severely debilitated individuals that may continue to inhale volatile agents, but respiration should be closely monitored for the occurrence of apnoea.

> Attempts at induction of anaesthesia in chelonian species using volatile agents alone are usually unsuccessful due to breath holding.

Injectable agents

A number of injectable agents can be used to sedate chelonia, either for minor procedures, such as venepuncture, or prior to induction with other agents. *Butorphanol* will lead to mild sedation, which may be sufficient to enable intravenous access to be gained for induction with propofol. *Ketamine* has been used to sedate red-eared sliders (*Trachemys scripta elegans*) both alone and in combination with xylazine or midazolam, but results were variable (Holz and Holz, 1994). Recovery times with ketamine are dose-dependent, with animals receiving anaesthetic doses taking more than 24 h to recover. Use of the tiletamine and zolazepam preparation in chelonia leads to prolonged recovery times (Schumacher and Yelen, 2006).

Sole use of *medetomidine* in chelonia is likely to cause severe cardiopulmonary depression, with reduced heart and respiratory rates and hypotension (Sleeman and Gaynor, 2000). Medetomidine can be used in combination with

Table 15.3: Sedative and anaesthetic agents used in chelonia (see also Table 12.6)

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Alfaxolone	10–15	IV	Smooth induction and recovery Light anaesthesia for <20 min
Alfaxalone/alphadolone	24[7]	IV	Muscle relaxation good, surgical anaesthesia (red-eared slider [*Trachemys scripta elegans*])
Atipamezole	5 × medetomidine dose[5]	IM	Reversal of medetomidine Severe hypotension if given IV (gopher tortoise [*Gopherus polyphemus*])
Butorphanol	0.2–0.4[8]	IM	Premedication: analgesia and sedation
Flumazenil	1 mg per 20 mg of zolazepam[17]	IM, IV	Reversal of zolazepam
Ketamine	20–60[4,9,10]	IM	Sedation Use low dose in debilitated animals
	60–90[10,13]		Light anaesthesia Prolonged recovery if high dose used
Ketamine + butorphanol	10–30 + 0.5–1.5[18]	IM	Deep sedation, minor surgery (e.g. shell repair)
Ketamine + midazolam	20–40 + 2[1]	IM	Sedation (in snapping turtles [*Chelydra serpentine*])
Ketamine + medetomidine	5 + 0.05[3]	IM, IV	Induction of anaesthesia (in loggerhead seaturtle [*Caretta caretta*])
	5 + 0.1[5]	IV	Short-term immobilisation for minor procedures (in gopher tortoise [*Gopherus polyphemus*]) Hypoxaemia and hypercapnia, moderate hypertension; advise oxygen supplementation and assist ventilation
Ketamine + midazolam	20–40 + <2[1]	IM	Sedation
	60–80 + < 2[13,18]		Anaesthesia
Medetomidine	0.15[20]	IM	Sedation (in desert tortoise [*Gopherus agassizii*]) with pronounced cardio-respiratory depression

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Midazolam	1.5[14]	IM	Sedation (study in red-eared sliders [*Trachemys scripta elegans*])
Neostigmine + glycopyrrolate	0.04 + 0.01[11]	IM	Reversal of rocuronium (glycopyrrolate administered 5 min prior to neostigmine, to antagonise possible parasympathetic effects of neostigmine)
Propofol	3–15[6,18,19,21]	IV	Induction of anaesthesia, titrate to effect, apnoea common
	1 mg/kg/min[18]		Continuous rate infusion to maintain anaesthesia
Rocuronium	0.25–5[11,12]	IM	Neuromuscular blockade, allowing intubation for anaesthesia with inhalational agents; require PPV until metabolised or reversed; no analgesia
			Recommend reverse at start of surgery when other anaesthetic agents employed, to allow monitoring of analgesia
Succinylcholine	0.25–1.0[2,15,16]	IM	Neuromuscular blocker; induction in 15–30 min
			No analgesia
			Not recommended in chelonian
Tiletamine/zolazepam	4–8[13]	IM	Sedation, may be sufficient to allow intubation
			Prolonged recovery (<72 h) at higher doses

Key: IM = intramuscular, IV = intravenous, PPV = positive pressure ventilation

[1] (Bienzle and Boyd, 1992); [2] (Boyer, 1992); [3] (Chittick et al., 2002); [4] (Crane et al., 1980); [5] (Dennis and Heard, 2002); [6] (Divers, 1996); [7] (Hackenbroich, 1999); [8] (Heard, 1993); [9] (Holz and Holz, 1994); [10] (Johnson, 1991); [11] (Kaufman et al., 2001); [12] (Kaufman et al., 2003); [13] (Millichamp, 1988); [14] (Oppenheim and Moon, 1995); [15] (Page, 1993); [16] (Page and Mautino, 1990); [17] (Raphael, 2003); [18] (Schumacher, 1996); [19] (Stahl and Donoghue, 1997); [20] (Sleeman and Gaynor, 2000); [21] (Wyneken et al., 2006)

ketamine, but oxygen and assisted ventilation should be administered to reduce the side effects, which may include hypoxaemia, hypercapnia and hypotension (Dennis and Heard, 2002). *Atipamezole* should not be administered intravenously in chelonia, as one study reported severe hypotension following dosing via this route (Dennis and Heard, 2002).

If venous access is available, the anaesthetic agent of choice for induction is *propofol*. Commonly the jugular vein is used for administration, but other options include the dorsal coccygeal vein, the subcarapacial vein or the intraosseous route. The dose is titrated to effect, as apnoea may occur with rapid administration of a bolus. Propofol is short acting and will allow either a short procedure to be performed, for example oesophagostomy tube placement, or intubation prior to maintenance with an inhalational anaesthetic agent for longer procedures.

Alfaxalone/alphadolone has been used in many chelonian species to induce and maintain anaesthesia. Induction is smooth and rapid, and recovery is usually 10–20 min after intravenous injection (McArthur, 2004a). It may also be administered intramuscularly, although response after intramuscular injection may be variable (Lawrence and Jackson, 1983; McArthur, 1996). Anaesthesia can be extended using top-ups without prolonging recovery. The author has also successfully used alfaxalone alone (Alfaxan®, Vétoquinol, Buckingham, UK) to induce anaesthesia in chelonia.

Anaesthetic maintenance

After induction of anaesthesia, most patients are intubated for provision of oxygen and inhalational anaesthetic agents. As the tracheal rings in chelonia are complete,

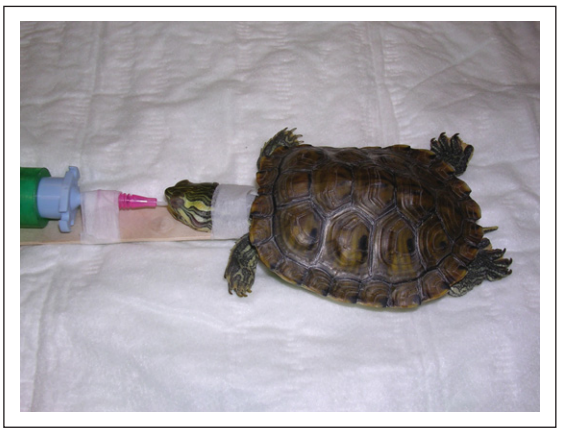


Figure 15.5 • Anaesthetised map turtle (*Graptemys* sp.) intubated with an intravenous cannula.

uncuffed endotracheal tubes are used. In cryptodirans, the tracheal bifurcation is quite proximal, and care should be taken not to intubate one bronchus (Schumacher and Yelen, 2006).

Owing to the respiratory depression caused by most anaesthetic agents, intermittent positive pressure ventilation (IPPV) is maintained throughout anaesthesia. If the patient is breathing voluntarily, this may be intermittent 'sighing' to ensure the lungs are well ventilated. If the patient's breathing is significantly depressed, including a decrease in tidal volume or a decrease in respiratory rate, continuous PPV should be performed. As discussed earlier (see Chapter 12), this may be carried out by an assistant, or using a mechanical ventilator. In either case, care should be taken not to over-inflate the delicate lungs, which may rupture. Observation of the normal degree of limb movement during inspiration is a useful guide to the pressure required. PPV is useful during coeliotomy, when the surgeon may benefit from knowledge of the timing of inspirations causing intracoelomic organ movement.

Recovery

Chelonian recovery from anaesthesia is similar to other reptiles. Special care must be taken with aquatic species not to return them to their aquarium until sufficiently recovered (usually 24 h after anaesthesia) or drowning may occur (Fig. 15.3). It is useful to use wet towels or incontinence pads along with regular spraying with water to keep the patient's skin moist during the recovery period, particularly in soft-shelled species.

Suggested protocols

Induction with intravenous propofol provides rapid and controllable induction. Without premedication, a comparatively high dose is required (up to 15 mg/kg). Alfaxolone, with or without alphadolone, may also be used intravenously to induce anaesthesia.

In patients where intravenous access is not possible, intramuscular agents are used. These may include ketamine, with or without medetomidine and/or butorphanol. Low doses will produce sedation to allow intravenous access for induction with propofol, while higher doses will induce anaesthesia. However, higher doses of intramuscular agents are often associated with prolonged recoveries.

After induction with the above regimes, the patient is then intubated and maintained on volatile agents with oxygen. Intermittent PPV is usually performed, either manually or with a mechanical ventilator.

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Crocodilian anaesthesia

16

INTRODUCTION

This section covers small crocodilians that may be kept by the reptile hobbyist. Local or national legislation may have specific requirements for the maintenance of these species in captivity, for example the Dangerous Wild Animals Act (1976) in the UK. All crocodilians should be considered dangerous, and due care taken when handling these species.

The three families within the Crocodilia order are Gavialidae (gharials and false gharials), Alligatoridae (alligators and caimans) and Crocodylidae (crocodiles). Although the larger representatives are unlikely to be presented to the general veterinary practitioner, smaller species, such as the dwarf caiman (*Paleosuchus palpebrosus*), may be seen.

ANATOMY AND PHYSIOLOGY

Temperature

As with other reptiles, environmental temperature is important in crocodilians, affecting behaviour and metabolism. Inadequate temperatures, below 25°C, will reduce appetite, digestion, and growth, and cause immunosuppression (Lane, 2006).

Cardiovascular system

The four-chambered crocodilian heart lies in the ventral midline, caudal to the forelimbs by approximately eleven scale rows (Hernandez-Divers, 2006). A small foramen, the foramen of Panizza, in the ventricular septum allows mixing of left and right ventricular blood, and the flow of blood to the respiratory system is reduced when the animal is submerged. When the animal re-surfaces, normal breathing causes a reduction in pulmonary pressures (and thence the right ventricle) that closes the foramen. Submerged crocodilians are commonly inactive and use anaerobic metabolism, allowing them to tolerate prolonged dives without re-breathing (Lang, 1987).

Respiratory system

The nostrils at the rostro-dorsal tip of the snout can be closed during submergence, and connect to the nasal passages. The gular fold or basihyoid plate (Fig. 16.1) prevents water entering the respiratory passages when the mouth opens underwater, for example when grabbing prey. The lungs are well developed, non-lobulated and highly vascular. The glottal valve retains air in the lungs for prolonged submergence (Lane, 2006). Crocodilians have a muscular pseudo-diaphragm.

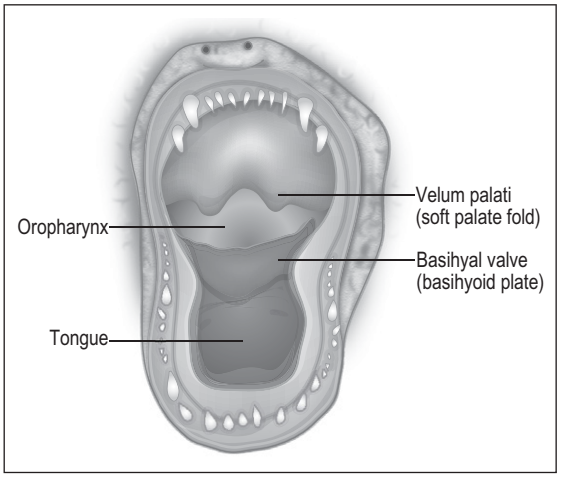


Figure 16.1 • Open-mouth view demonstrating the gular fold or basihyoid valve in crocodilia.

Digestive system

This group are carnivorous, eating both live prey and carrion in the wild. They will eat at temperatures between 25°C and 35°C (Lane, 2006).

Integumentary system

The skin has separate scales joined by connective tissue. Crocodilians have dorsal cornifications over bony plates, which precludes injections in this region (Lane, 2006).

- Low temperatures produce immune suppression.
- The crocodilian heart has four chambers (unlike other reptiles that have three).
- The gular fold allows breathing through the nose while the mouth is open under water.
- Crocodilians have a muscular pseudo-diaphragm.
- Bony plates dorsally and thickened skin make injections difficult.

TECHNIQUES

Restraint

Initial restraint should protect handlers from bites and scratches. The snout should be taped shut; gloves can be worn and towels used during restraint to reduce trauma from sharp claws. Several personnel will be required to restrain larger specimens.

Routes of administration

These sites are similar to those used in lizards. The main difference is the use of larger and longer needles to penetrate the skin for injections.

Oral

Owing to staff safety issues, the oral route is used to administer medication only in very small or severely debilitated animals. Medications can be added to feed. In very small animals, gavage dosing is similar to lizards, except with the mouth taped shut once a gag has been placed. The dosing tube must pass over the basihyal valve in the pharynx (Mitchell, 2006).

Subcutaneous

This is rarely used in crocodilians due to their thick dermal scales.

Intravenous

The easiest site for venous access is the ventral coccygeal vein in small or medium animals and the supravertebral vein in medium or large animals. The ventral coccygeal vein is accessed as for lizards. The supravertebral vein is accessed in the dorsal midline just caudal to the occiput, with the needle perpendicular to the skin. Slow advancement will avoid spinal trauma (Hernandez-Divers, 2006). In an emergency, the heart can be injected, but cannot be stabilised (Fudge, 2000).

Intubation

After induction, the jaws are held open and the gular fold (see Fig. 16.1) deflected to visualise the glottis for intubation.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Owing to the demeanour of these species, most anaesthetic protocols are based on injectable agents. There is wide variation in species response to different agents. Muscle relaxants are commonly used to assist immobilisation, but do not provide analgesia. Induction with intravenous propofol into the ventral coccygeal vein may be possible in small specimens.

Species variations in response to anaesthetic agents exist. For example, alligators respond well to etorphine, which is not very effective in other species. Gallamine, on the other hand, appears to be unsafe in alligators at doses greater than 1 mg/kg (Page, 1993). Ketamine may be used to sedate or anaesthetise crocodilians; recovery times are dose-dependent and may be prolonged.

The intramuscular or subcutaneous routes are usually used to induce anaesthesia in crocodilian species. Addition of hyaluronidase to the anaesthetic agent accelerates absorption (Lloyd, 1999).

Neuromuscular blockers

Crocodilians are a species in which neuromuscular blockers are commonly used. The agent produces immobilisation, enabling safer handling and induction of anaesthesia for painful procedures. These drugs do not produce analgesia. Intermittent positive pressure ventilation (IPPV) should be performed in animals that have received these agents.

Anaesthetic maintenance

If possible, the patient is intubated and maintained on volatile agents. Some injectable regimes will produce anaesthesia, but oxygen should still be supplemented if available.

Table 16.1: Anaesthetic agents used in crocodilians (see also Table 12.6)

DRUG	DOSE (mg/kg)	ROUTE	COMMENT
Alpha-2-agonists:			
Medetomidine	0.15[9]	IM	Sedation (incomplete immobilisation)
Xylazine			Bradycardia and bradypnoea seen
	1–2[5,7]	IM	Preanaesthetic (study in Nile crocodiles)
Benzodiazepine:			
Zolazepam	See tiletamine combination below	–	–
Reversal agent:			
Flumazenil	1 mg per 20 mg zolazepam[5]	IM, IV	Reversal of zolazepam
Dissociative agent:			
Ketamine[5]		SC, IM, ICe	Induction <30–60 min, recovery in hours to days
	20–40		Sedation
	40–80		Anaesthesia
Dissociative agent combinations:			
Ketamine + medetomidine	5–15 + 0.1–0.25[2]	IM	Anaesthesia (in American alligators [*Alligator mississippiensis*])
			Lower doses required for adults compared to juveniles
			Reverse medetomidine with atipamezole
Tiletamine/zolazepam	5–15[1,8]	IM	Immobilisation for restraint and intubation
			Recovery may be prolonged
Neuromuscular blockers:[9]			Can be used alone or with ketamine or benzodiazepines to provide immobilisation
			No analgesia
			Use lower dose in larger animals
Gallamine	0.5–2[3]	IM	–
Succinylcholine	0.25–1.2[3]	IM	Variable induction and recovery periods with succinylcholine
Reversal agent:			
Neostigmine	0.03–0.06[3]	IM	Antidote to gallamine (side effects include emesis and lacrimation, so often allow recovery without reversal)
			Can combine with hyaluronidase (75 mg) to enhance effects[6]
Opioids:			
Etorphine HCl[8]	0.30–2.75[4]	IM	Crocodilians require high doses
			Poor relaxation
			Legal restrictions due to safety issues
Propofol	10–15[5]	IV	Anaesthesia 30–90 min
Hyaluronidase	25 IU/dose[5]	SC	Combine with other drugs to accelerate SC absorption

Key: ICe = intracoelomic, IM = intramuscular, IV = intravenous, SC = subcutaneous

[1] (Clyde et al., 1994); [2] (Heaton-Jones et al., 2002); [3] (Lane, 2006); [4] (Lawton, 1992); [5] (Lloyd, 1999); [6] (Lloyd et al., 1994); [7] (Page, 1993); [8] (Schumacher and Yelen, 2006); [9] (Smith et al., 1998)

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17 Amphibian anaesthesia

INTRODUCTION

Over 4000 species belong to the class Amphibia, in three orders: Anura (Salientia) – frogs and toads; Caudata (Urodela) – salamanders, newts and sirens; and Gymnophiona (Apoda) – caecilians (Adler, 2004). Many species of amphibian are endangered in the wild, mostly due to environmental factors, such as pollutants and loss of breeding sites, or fatal infections, such as chytridiomycosis (Berger et al., 1999; Daszak et al., 1999).

Members of the order Anura are most often seen as pets, with common species kept in captivity including White's treefrog (*Pelodryas caerulea*) and poison arrow frogs (*Dendrobates* spp.). Caudata are seen less frequently, for example fire salamander (*Salamandra salamandra*) (Fig. 17.1). Gymnophiona are uncommonly presented at veterinary practices. In the following discussions, salamanders and caecilians will be discussed as the predominant members of the Caudata and Gymnophiona, respectively.

Chemical restraint may be necessary for several reasons. Difficulties may be associated with conscious examination, particularly with smaller species (for example, the spot-legged poison frog [*Epipedobates pictus*] may die after a few



Figure 17.1 • Fire salamanders, *Salamandra salamandra*.

Table 17.1: Amphibians commonly kept as pets

COMMON NAME	LATIN NAME	ORDER	COMMENT
African bullfrog	*Pyxicephalus adspersus*	Anura	–
African clawed frog	*Xenopus laevis*	Anura	Completely aquatic; common in laboratories
Fire-bellied toad	*Bombina* sp.	Anura	–
Green tree frog	*Hyla cinerea*	Anura	–
Leopard frog	*Rana pipiens*	Anura	–
Cane (marine) toad	*Bufo marinus*	Anura	–
Poison dart frog	*Dendrobates* sp.	Anura	Require high humidity; arboreal; parental care of tadpoles
White's tree frog	*Litoria caerulea*	Anura	–
Mexican axolotl	*Ambystoma mexicanum*	Caudata	Neotenic
Mudpuppy	*Necturus maculosus*	Caudata	Obligate aquatic
Tiger salamander	*Ambystoma tigrinum*	Caudata	–

(Raphael, 1993, Wright, 2001e)

minutes of manual restraint) or more aggressive species (for example, horned frogs [*Ceratophrys* spp.] may bite when provoked) (Buchanan and Jaeger, 1995; Wright, 2001c). Internal transillumination (a useful technique for visualisation of intracoelomic organs) and some sampling techniques (for example, blood sampling using venepuncture or cardiocentesis, tracheal swab) may require sedation or general anaesthesia. Analgesia and anaesthesia will be required if surgery is to be performed in these species.

ANATOMY AND PHYSIOLOGY

Temperature

Amphibians are ectotherms and, consequently, care should be taken to work in an appropriate environmental temperature throughout the procedure. The preferred body temperature will vary according to species, age, season and current metabolic processes (Goin et al., 1978; Whitaker et al., 1999; Wright, 2001b). Changing temperatures will affect blood gas composition as well as acid–base status (Boutilier et al., 1987). The optimal temperature for temperate frogs will be 20–25°C compared with 25–30°C for tropical species; 10–16°C is appropriate for temperate salamanders and 15–20°C for tropical species (Jaeger, 1991; Raphael, 1993). Lower temperatures (0–20°C) may cause long-term problems with immune suppression, while higher temperatures enhance the immune response. Many infections in captive amphibians originate as ubiquitous bacteria or fungi, which opportunistically cause infection in immunosuppressed individuals.

Water quality

Aquatic species and larval forms require water for immersion. The water should be dechlorinated; this is achieved by standing the water at room temperature, adding dechlorination tablets, or by charcoal filtration. Optimal pH will vary between species and developmental stage, but is usually between 6.5 and 9.5. Water cleanliness is performed by filtration as in fish vivaria or by regular replacement of portions of the water (Raphael, 1993).

In terrestrial species, environmental humidity is important. Temperature species should be maintained in 60–80% humidity, and tropical species in 70–90%.

Cardiovascular system

This includes the arterial, venous and lymphatic systems. Lymph contains all the components of blood except for erythrocytes. The lymphatic systems contain lymph sacs (also known as lymph hearts or lymph vesicles) that restrict lymph flow to unidirectional. These structures beat in synchrony independent of the cardiac rate, at approximately 50–60 beats per minute (Conklin, 1930). Well-hydrated terrestrial amphibians may absorb fluid from the skin directly into lymph (Boutilier et al., 1992). This fluid is eliminated by the kidneys, which may affect the distribution and pharmacokinetics of substances absorbed across the skin (Wright, 2001a).

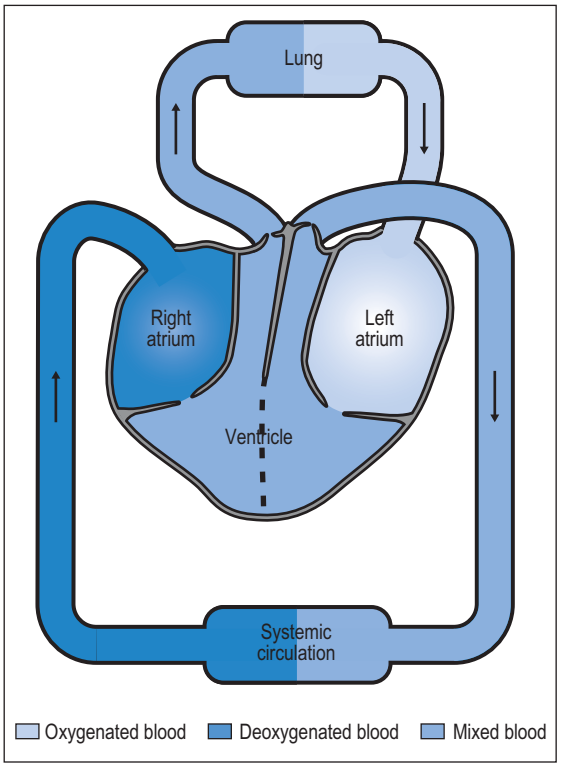


Figure 17.2 • Schematic showing the amphibian circulation.

The typical amphibian heart is three-chambered, with a right atrium, a left atrium and a single ventricle (Fig. 17.2). Species differences occur (see below), such as septal fenestrations that allow mixing of oxygenated and deoxygenated blood (Wallace et al., 1991). As with reptiles, blood flow within the heart may vary during a dive. For example, in the African clawed frog (*Xenopus laevis*) ventricular blood flow is separated during air breathing, but less divided when the animal dives (Emilio and Shelton, 1974). Central blood pressures between various anuran species appear to be similar, but are significantly lower in *Salamandra* species (Shelton and Jones, 1968).

Renal and hepatic portal vein systems exist in the caudal amphibian. Little pharmacokinetic data are available for amphibian species and so drugs that may be metabolised or excreted renally or hepatically should not be administered in the hindlimbs.

The adult *anuran* heart is dorsal to the pectoral girdle and sternum (Figs 17.3 and 17.4). The inter-atrial septum is complete and ventricular trabeculae are present in some species (Kumar, 1975). The presence of a large lingual venous plexus (Fig. 17.5) is useful for venepuncture, including administration of drugs during anaesthesia. Other options for venepuncture include the midline abdominal vein (which may be visible percutaneously in

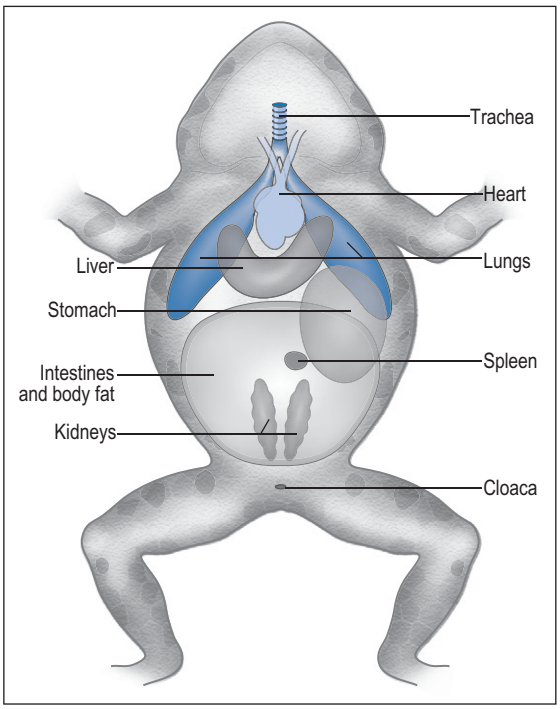


Figure 17.3 • Schematic to show location of major organs in an anuran (ventral view).

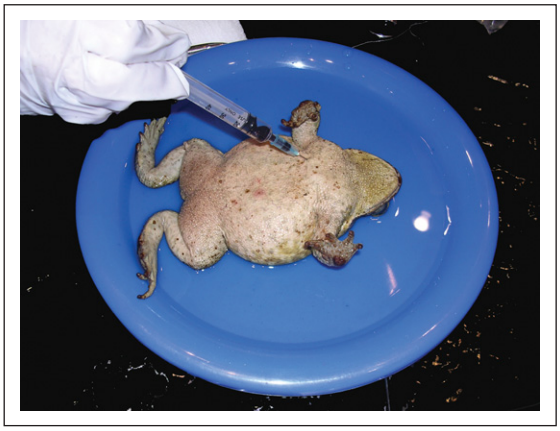


Figure 17.4 • Cardiocentesis in an anaesthetised anuran (cane toad, *Bufo marinus*).

frogs with pale-coloured ventral skin) (Fig. 17.6), heart (Fig. 17.4) and femoral vein (in larger species).

Anurans possess few lymph hearts compared to other amphibians. A prominent pair may be present bilaterally dorsally at the urostyle in terrestrial species, from which lymph may be collected or fluids administered (Fig. 17.7) (Carter, 1979).

Adult *salamander* cardiac anatomy varies greatly between species. A ventricular septum is present in the lesser siren



Figure 17.5 • The lingual venous plexus is located by pulling the anuran (in this case a cane toad [*Bufo marinus*]) tongue forward, and can be utilised for venous access.

(*Siren intermedia*) and mud-puppy (*Necturus maculosus*) (Putnam, 1975; Putnam and Dunn, 1978). The interatrial septum in salamanders is fenestrated (exceptions are sirens [*Siren* sp.] and the hellbender [*Cryptobranchus alleganiensis*]). The interatrial septum is modified in Plethodontidae (lungless salamanders) (Putnam and Kelly, 1978). Again, the midline abdominal vein is prominent, but the ventral tail vein may be a more easily accessible site for venepuncture in salamanders.

The atrial septum is fenestrated in adult *caecilians*, and the left atrium is usually smaller than the right. Caecilians have over 200 lymph hearts subcutaneously (Wright, 2001a).

Respiratory system

The amphibian respiratory system varies between orders and individual species, and even intraspecies depending on the individual's life-stage and environment. Most larval stages are aquatic and rely on branchial respiration, having external gills.

The anuran tongue is attached rostrally and at rest lies pointing caudally within the oral cavity. The glottis is at the base of the tongue, on the ventral buccopharyngeal cavity. In most amphibian species, the trachea is short (Fig. 17.3), with cartilaginous rings. The trachea birfurcates into pulmonary bronchi.

Many immature amphibians are totally aquatic and rely on branchial respiration. External gills are often lost when metamorphosis occurs, for example when tadpoles become froglets.

Respiration in adult *anurans* may be cutaneous, buccopharyngeal and/or pulmonic. Adaptations include an increase in surface area via skin folds in the aquatic Titicaca water frog (*Telmatobius coleus*), which relies mostly on cutaneous respiration, living in a cold well-oxygenated mountain lake. By contrast, the African clawed frog (*Xenopus laevis*) is primarily a lung breather, as adequate cutaneous respiration is not possible in the poorly oxygenated warm water in which it lives (Wright, 2001a). African clawed frogs with access to air will consume approximately one-quarter of their oxygen via the skin (Emilio and Shelton, 1974).

Mechanisms for buccal ventilation differ between anurans (Brett and Shelton, 1979). The left and right anuran lungs are approximately the same size and are simple structures without partitioning or infolding (Wright, 2001a).

Salamander respiration may be branchial, cutaneous, buccopharyngeal or pulmonic. Neotenic species (for example, axolotls [*Ambystoma mexicanum*]) have external gills and rely heavily on branchial respiration (Fig. 17.8). Some aquatic species (for example, sirens [*Siren* spp.]) have lungs as well as gills. Gill structure varies, depending on the species and environment (especially oxygen content and speed of water flow). All salamander species perform cutaneous respiration to variable degrees, with gaseous exchange across the skin promoted by several features: a thin epidermis, highly vascular dermis, high surface area to volume ratio and cylindrical shape. A low metabolic rate in these species and ability to incur an oxygen debt via anaerobic glycolysis also aid this form of respiration (Wright, 2001a).

Lung size and partitioning vary between salamander species. The left lung is slightly larger than the right. In aquatic salamanders, the simple lungs are more caudally located, and consequently the trachea is longer. The lungs in terrestrial species usually have sacculations and some

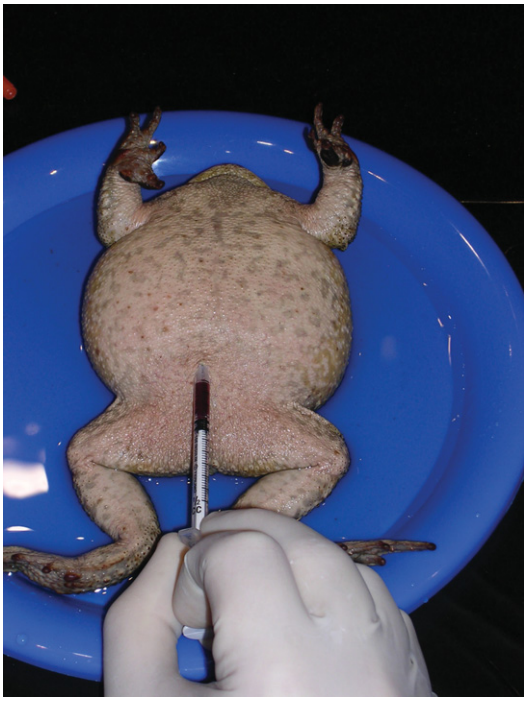


Figure 17.6 • Venepuncture is possible in the midline ventral abdominal vein (shown in an anaesthetised cane toad [*Bufo marinus*]).

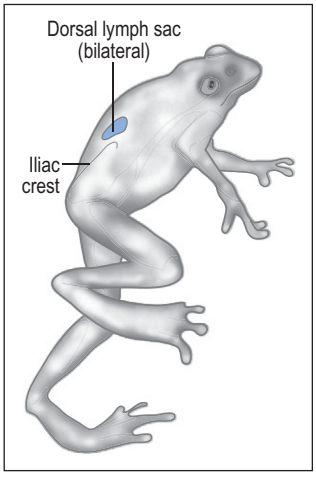


Figure 17.7 • Location of dorsal lymph sacs in anurans.

Table 17.2 Respiration in adult amphibians (species variations exist, and not all species will show all methods of respiration)

	CUTANEOUS	BRANCHIAL (GILLS)	BUCCOPHARYNGEAL	PULMONIC
Anurans	Yes	No	Yes	Yes
Caudata	Yes	Yes	Yes	Yes
Gymnophiona	Yes	No	Yes	Yes

alveoli, as do some pond-dwelling species. Some species have reduced or absent lungs (Wright, 2001a).

Adult *caecilians* have pulmonic, buccopharyngeal and cutaneous respiration. The primary mode will vary between species, with the lungs being most important (Bennet and Wake, 1974). The left lung is often smaller than the right or absent. The lung lobes are elongate, with alveoli present. Smooth muscle controls the diameter of the external nares and choana. The buccopharyngeal structures are primarily responsible for inspiratory and expiratory effort (Mendes, 1945). A tracheal lung with respiratory epithelium exists in some species of ichthyophiids (fish caecilians) and typhlonectids (aquatic caecilians) (Wright, 2001a).

The drive for respiration in amphibians may be hypoxia and/or hypercapnia; species differences exist (West and Van Vliet, 1992). Stretch receptors in the lungs and chemoreceptors in lungs or blood appear to be involved in vasodilation of the pulmonary vascular bed during breathing (Emilio and Shelton, 1972). These changes are mediated in the central nervous system and result in an increase in pulmonary blood flow and a decrease in pulmocutaneous arterial pressure. This vasodilation response occurs as part of normal diving-emergence behaviour in toads.

Metabolism

Normal energy production is via aerobic metabolism, but anaerobic pathways are employed during outbursts of activity (Gatten et al., 1992; Pough et al., 1992).

Urinary system

Amphibian kidneys are mesonephric and so cannot concentrate urine higher than the concentration of the solutes in plasma. The kidneys filter vascular and coelomic fluid, which may include intracoelomically administered medications (Wright, 2001a).

In *anurans*, the primary nitrogenous waste product depends on the species' environment. In aquatic amphibians, ammonia is produced and freely diffused into the environment across the skin and via renal excretion. Similar to other aquatic animals, they have little concentrating ability. In these species, the liver plays a small role in nitrogen processing. In terrestrial amphibians, nitrogenous waste is primarily excreted as urea, with conversion of ammonia to urea (and to uric acid, to conserve water in a small number of species) occurring in the liver. A few terrestrial species in drier conditions convert nitrogen wastes to uric acid to conserve water further (Wright, 2001a).

Secondary tubules are found in the mesonephric kidney of most species of adult *salamander*. In many species the emphasis for nitrogenous waste excretion is on vascular filtration. Again, renal relevance is dependant on lifestyle and environment. The excretory requirements of *caecilians* may change from larva to adult (increasing for terrestrial species).



Figure 17.8 • Axolotl, *Ambystoma mexicanum*, showing external gills in this neotenic species.

Digestive system

Amphibians are carnivorous, with most being insectivorous, and require a high protein diet. The exact diet will depend on the species, but is quite varied in the wild. Smaller amphibians usually eat invertebrates while larger species may consume vertebrates (Adler, 2004). In captivity, many are fed live items, such as crickets, mealworms, tubifex worms, mice pups and fish. Dry dog, cat and fish food may also be used. Secondary nutritional hyperparathyroidism is common in captive amphibians and may predispose to muscle fasciculations, seizures or pathological fractures. Feeding a variety of foods will help prevent nutritional deficiencies and vitamin/mineral supplements are usually added to invertebrate diets. Vitamin C is essential in tadpole diets (Raphael, 1993).

Diet may be affected by life-stage and many tadpoles are herbivorous, converting to carnivory or insectivory after metamorphosis. Day/night cycles may affect metabolism. The time of day for feeding can, therefore, affect digestion and absorption.

Integumentary system

Amphibian skin is important for thermoregulation and hydrational homeostasis (Heatwole and Barthalamus, 1994). The highly vascular and well-innervated dermis is covered by the epidermal layer that is several cell layers thick, but thinner than in other tetrapods. The stratum corneum is only a single layer thick or lacking altogether in some aquatic salamanders. Most amphibians do not have dermal scales (the exception being some caecilians) (Helmer and Whiteside, 2005). Anurans lack tight collagen fibres between the stratrum spongiosum and underlying tissues, creating a potential subcutaneous space for fluid administration (Wright, 2001a). Subcutaneous fluid accumulation may be normal in these species.

The thin epidermis is easily torn and defects will allow entry of pathogenic organisms. Moistened latex or nitrile gloves worn by the clinician will help protect this layer during restraint. To protect the amphibian from noxious substances, any lubricating powders should be rinsed off

gloves prior to handling the animal. Water-soluble non-toxic gel may also be applied, for example when handling delicate skinned aquatic species (some of which do not possess keratinisation of the stratum corneum).

An amphibian's skin is of paramount importance for water conservation. The skin of leaf frogs (*Phyllomedusa* spp.) secretes a lipid substance as waterproofing and is particularly resistant to evaporative water loss (Blaylock et al., 1976; McClanahan et al., 1978). However, for most amphibians, the skin is a negligible barrier to water loss. As mentioned above, most species rely heavily on cutaneous respiration and their skin must be kept moist for gas exchange. An area on the ventrum, the 'drinking patch', is responsible for most water uptake (Parsons, 1994).

Care should be taken when handling amphibian species. Many produce skin secretions: glue-like mucilaginous secretions (for example, slimy salamanders [*Plethodon glutinosus*]) or malodorous secretions (for example, mink frog [*Rana septentrionalis*]) (Chen and Chen, 1933; Wright, 2001c). Some of these secretions will cause a variety of effects in humans: mild-to-severe inflammation of mucous membranes (for example, Bufonidae spp. toads); and salivation, regurgitation, dyspnoea, convulsions and even death (for example, the cane toad [*Bufo marinus*] or poison arrow frogs [*Dendrobates* spp.]). Secretions are often increased when the animal is injured or agitated. Species such as the spotted toad (*Bufo guttatus*) can expel secretions from their parotid glands over long distances (up to 2 m). For these animals, caution should be exercised and further personal protective equipment (for example, goggles) may be necessary. Catecholamine release during restraint or other stress can also influence skin colour change (Raphael, 1993). It is prudent to wear protective gloves when handling amphibians, either conscious or anaesthetised.

BOX 17.1 Important points: anatomy and physiology

Heart
- Three-chambered – right atrium, left atrium, single ventricle

Respiration
- Cutaneous, branchial (gills), buccopharyngeal, pulmonic

Urinary
- Cannot concentrate urine higher than plasma solute concentration
- Main nitrogenous waste depends on species

Skin
- Easily damaged
- Important for thermoregulation and water homeostasis.

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

Before embarking on anaesthesia of a patient, a complete history should be taken and physical examination performed.

History

The history should include signalment (species, age, sex if known) along with both previous and current husbandry and medical problems (including treatments administered). Analysis should be performed on a water sample from the amphibian's enclosure to ascertain water quality.

Clinical examination

As with many pet species, zoonotic diseases are potentially present and the clinician should exercise good personal hygiene during the examination. Immunosuppressed amphibians in particular may suffer from infections, such as mycobacteria and *Chlamydophila psittaci*.

The patient is observed within the enclosure to assess skin colour, body condition, respiratory rate, demeanour and activity. A brief clinical examination should be possible in most conscious patients (Wright, 2001c). Clinicians should wear gloves to protect themselves from toxins in the amphibian's skin, rinsing powder off the gloves beforehand, to reduce damage to the patient's delicate skin. The animal should be examined quickly to identify gross abnormalities, including assessment of the oropharynx, nares, eyes and skin. The heartbeat can be observed on the ventral body surface (see Fig. 17.4). As with some reptile species autotomy is possible in some salamanders and the owner should be warned of the risk of tail loss during restraint. If more prolonged investigation is required, the patient should be remoistened in a container of water to prevent desiccation during the procedure (Raphael, 1993).

Poor husbandry conditions often contribute to ill health in amphibian pets. Any anomalies will predispose the patient to disease, which may be clinical or sub-clinical. Infections are often opportunistic in immunocompromised individuals. In these cases, elective procedures may be postponed until husbandry corrections have been implemented or critical patients stabilised (for example, with appropriate husbandry and/or fluid administration) for a few days prior to anaesthesia. Chronic disease processes, such as renal disease associated with dehydration, will greatly increase the anaesthetic risk. In certain cases, anaesthesia cannot be postponed and in these cases owners should be made aware of the risks involved. Peri-anaesthetic care and monitoring can reduce these risks.

Hospitalisation

The basic amphibian hospital enclosure comprises a glass or Perspex tank. Moistened paper towels can be used as

substrate, an upturned plastic cup for shelter and a shallow water dish should be provided. Aquatic species need to be able to immerse themselves in fresh chlorine-free water. Other important environmental conditions include ultraviolet (UV)-B lighting and temperature in an appropriate range (see Anatomy and physiology section), and relative humidity at 70–80% (Hadfield and Whitaker, 2005).

Water quality prior to, during and post anaesthesia is particularly important for aquatic species. For these species in constant intimate contact with water and for all larval forms, dechlorinated water should be used for procedures. The pH should also be monitored closely, as changes will affect the sensitive skin.

Fluid therapy

Maintenance of water and electrolyte balance is vital for amphibians. Amphibian patients may present with dehydration or may become dehydrated during procedures at the veterinary surgery. Water has much lower osmolality (20–40 mOsm for 'fresh' water) than plasma in a healthy individual (usually at least 250 mOsm) (Boutilier et al., 1992). Animals spending time in water may develop imbalances leading to skin, gill or renal damage. Damaged epithelia may then lead to electrolyte loss and water influx may be excessive. Fluid overload may occur if ill animals are maintained in 'fresh' water. Conversely, dehydration (including electrolyte loss) may occur in terrestrial species and should also be prevented or corrected.

In either of these situations, *bathing* the patient in a shallow electrolyte bath is a simple method of treatment of dehydration or as prevention of imbalance in an ill animal (for example, amphibian Ringer's solution or Whitaker–Wright solution at various concentrations [Table 17.3]).

Balanced electrolyte solutions may be administered *intracoelomically*. However, many solutions for mammals are hypertonic for amphibians or contain excess lactate (which may cause lactic acid overload in the amphibian). One or two parts balanced electrolyte intravenous solution added to one part 5% dextrose is more suitable for intracoelomic administration in dehydrated amphibians. In general, topical fluid therapy via baths is more applicable to maintenance of amphibian water and electrolyte balance (Wright and Whitaker, 2001). *Intravenous* access may be possible in larger species. A maximum volume of 25 ml fluid per kilogram of body weight should be administered parenterally (Wright, 1996).

Nutritional support

Carnivore convalescent diets (for example, Hills® a/d, Herts., UK) can be administered to support adult amphibians. Initially, 1% of body weight can be administered once daily diluted in a similar volume of chlorine-free fresh water. This volume can be gradually doubled (Hadfield and Whitaker, 2005). Body condition should be assessed with daily weight checks.

Table 17.3: Electrolyte baths for amphibians

Amphibian Ringer's solution (Humason 1967, as cited in (Maruska, 1994))

Distilled water	1 L
NaCl	7.3 g
KCl	0.17 g
$CaCl_2$	0.17 g
$NaHCO_3$	0.22 g

- Mix to dissolve all crystals; agitate before use. Keep in closed container to reduce evaporation. Use 10–100% solution for ill aquatic amphibian

Whitaker–Wright solution (100% solution) (Wright and Whitaker, 2001)

Distilled water	1 L
NaCl	113.0 g
$MgSO_4 \cdot H_2O$	8.6 g
$CaCl_2$	4.2 g
KCl	1.7 g

- Dissolve crystals thoroughly
  Cover container to prevent evaporation. Add Trizma® buffering crystals (Sigma, St Louis, Missouri) to stabilise between pH 7.0 and 7.3
- Dilutions can be made from this solution. e.g., 5 ml of 100% solution with 95 ml distilled water will make 5% solution; at 21.7°C and pH 7.0 this will have an osmolality of 206 mmol/kg.
  Use 5% solution if no fluid overload is present, or 10% if severe fluid overload is present

Saline (Wright and Whitaker, 2001)

- 0.8–2.5%: Hypertonic saline may be used if solutions above are not available, for a maximum of 4 h
- 0.5–0.8%: Normal saline is suitable as a bath for ill terrestrial amphibians. (Provide a bath of untreated fresh water simultaneously)

EQUIPMENT REQUIRED

Water should be clean and chlorine-free. It is important that it is also at the correct temperature and pH (particularly if the anaesthetic agent used requires buffering) and well oxygenated. Separate buckets of fresh water should be available for recovery, along with the anaesthetic solution for induction and a less concentrated anaesthetic solution for maintenance.

Plastic bags are useful for induction, as the amphibian is less likely to traumatise itself than in solid containers. Syringes are used to squirt liquid over the skin to moisten it during the procedure. A waterproof tank and/or a shallow bowl or plate serves as a maintenance platform.

Anaesthesia monitoring is usually carried out using a Doppler probe with a small footprint.

TECHNIQUES

Routes of administration

Injections

Prior to administration of injections, the skin should be wiped clean of debris and excess skin secretions. Sterile saline may be used. Care should be taken with disinfection. Isopropyl alcohol is toxic in amphibians (Wright, 2001d). Amphibians are also susceptible to water-soluble toxins such as iodine, but safe use of tamed-iodine compounds has been documented (Brown, 1995; Jacobson, 1975). Dilute chlorhexidine (1:40) is appropriate for use on amphibian skin (Whitaker and Wright, 2001).

For *intracoelomic* injections, the patient is held in dorsal recumbency with the caudal end slightly elevated, in an attempt to avoid organ puncture. A small gauge needle (23-gauge or smaller) is used, inserting in a paramedian or paralumbar location in the right posterior quadrant of the abdomen (Lewbart, 2001).

The skin of some species contains ossifications, which may make injections difficult (Wright, 2001a).

Anurans can also be injected *subcutaneously* as their skin adheres loosely to underlying tissues (see Anatomy section), but not salamanders and caecilians. Fluids or some medications can be administered into *lymph sacs*, for example the dorsal lymph sacs of anurans, which are cranio-lateral to the urostyle (see Fig. 17.7).

In larger amphibians, *intramuscular* injections can be administered, for example into the proximal limbs. The posterior muscles of the forelimb or hindlimb are used in salamanders, and the hindlimb of frogs (Lewbart, 2001).

Intravenous injections are difficult, but possible in larger animals. The ventral abdominal vein lies just dorsal to the skin (see Fig. 17.6), and the dorsally recumbent patient is approached with the needle bevel uppermost directed at an approximately 45° angle. In order to access the lingual plexus, which lies bilaterally at the base of the tongue, the tongue must be pulled rostrally out of the mouth (see Fig. 17.5). The heart lies midline in the pectoral region and is located by visualisation, palpation or Doppler flow. It is accessed by directing a needle almost perpendicular to the skin just to the right of midline (see Fig. 17.4). (Lewbart and Stoskopf, 2002). The femoral vein may be accessible in some larger species. The ventral tail vein is the most easily accessible vein in salamanders (Whitaker and Wright, 2001).

Intubation

Most animals will not require intubation, as cutaneous respiration is relatively efficient. The anuran tongue is flipped forward to access either the glottis for intubation or the lingual venous plexus for venepuncture (see Fig. 17.5).

Large specimens can be intubated using small endotracheal tubes or flexible intravenous catheters. The amphibian trachea is short and the endotracheal tube should not be inserted too far (Raphael, 1993). The trachea is also narrow in some aquatic species compared to a similar-sized terrestrial amphibian.

It can be difficult to maintain anaesthesia in amphibians via this method due to the alternative respiratory methods that most species possess.

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Induction

Immersion

This is the most common method of anaesthetising amphibians. The technique is described in detail in the section below, but can be applied to other agents.

Tricaine methanesulfonate (MS-222)

The agent of choice for amphibian anaesthesia is MS-222 (PHARMAQ Ltd, Hampshire, UK; Finquel®, Argent Chemical Laboratories, Redmond, WA). MS-222 is a sodium-channel blocking local anaesthetic. It is an isomer of benzocaine and is a water-soluble white salt that forms a clear, colourless, acid solution when mixed with water. Anaesthesia is induced by bathing the amphibian in the solution (Raphael, 1993). The solution has a wide safety margin, with the LC_{50} in frog toxicity studies being 6.2% (Finquel® data sheet, Argent Chemical Laboratories). MS-222 directly affects the respiratory motor output from the central nervous system, causing both excitation and inhibition (Hedrick and Winmill, 2003). Prolonged exposure may affect renal circulation and cause deaths (Finquel® data sheet, Argent Chemical Laboratories).

Care should be taken to avoid human exposure (skin or eye contact, inhalation or ingestion) to either the MS-222 powder or solutions, as they can be irritant or corrosive. Solutions should not be disposed of into water supplies or natural waters.

MS-222 is supplied as an acid salt. In solution (in its un-ionised or free base form), this agent can be used to produce anaesthesia in amphibians. However, when MS-222 is added to water the solution formed is acidic and mostly contains ionised MS-222 (which cannot be absorbed across amphibian skin). Therefore, the solution must be buffered before use. If the MS-222 is not buffered, iatrogenic metabolic acidosis may be induced (Hadfield and Whitaker, 2005).

Sodium phosphate (Na_2HPO_4) is the easiest buffer to use, as excess will not affect the ratio of ionised to unionised MS-222. Addition of MS-222 to a solution of excess phosphate buffer produces a pH of around 7. Alternatively the agent can be free-based by the addition of sodium bicarbonate ($NaHCO_3$, bicarbonate of soda) to a solution of the MS-222 salt, whilst monitoring the pH

to achieve a point around pH 7–7.4. Excess sodium bicarbonate will result in a pH outside the physiological range. It will also increase the concentration of un-ionised MS-222. The pH of local water will affect the amount of buffer required – this will be more critical when using sodium bicarbonate.

To reduce stress caused to the animal by sudden changes in water parameters, the patient's enclosure

BOX 17.2 Tricaine methanesulfonate (MS-222) 0.1% solution, 1 g MS-222/L (Crawshaw, 1992)

2 g MS-222 + 34–50 ml of 0.5M Na_2HPO_4 (buffer) + 2 L of oxygenated water

pH = 7.0–7.4

Dilute as required with well-oxygenated, toxin-free water

water is used to prepare the anaesthetic solution if it is of sufficient quality. Toxin-free water with buffering agent can be stored and MS-222 added when anaesthetic solution is required. It is useful to have water available in measured (for example 1 L) quantities for further dilution of the anaesthetic agent or for recovery. All solutions should be allowed to equilibrate to room temperature, to reduce thermal shock caused to the animal due to temperature changes (Wright, 2001c).

A wide range of MS-222 concentrations are reported to be effective in anaesthetising amphibians (Crawshaw, 1989; Vanable Jr, 1985). These will vary between species and between life stages. Recommended induction concentrations are 0.2–0.5 g/L for larvae and newts, 1–2 g/L for adult frogs and salamanders, and 3 g/L for toads (Crawshaw, 1993; Downes, 1995; Hadfield and Whitaker, 2005). Large species (for example, the Japanese giant salamander [*Andrias japonicus*] can weigh over 60 kg) may require higher concentrations for induction, but care should be taken to buffer the solution appropriately.

It may, therefore, be necessary to titrate the dose for different species and individual animals. In general, the concentration of MS-222 necessary and time taken to reach surgical anaesthesia is longer in toads due to slower absorption of fluid and drugs across the skin compared to frogs.

The patient is bathed in the anaesthetic solution in either a plastic 'zip-lock' bag (Fig. 17.9) or a lidded plastic container. The patient can be protected from self-trauma in the latter by lining it with a cloth. On initial exposure to MS-222, erythema is often seen (more obviously on the pale-skinned ventrum), followed by agitation and, finally, loss of the righting reflex. Most patients will require approximately 5 min in contact with MS-222 solution before becoming anaesthetised, with the resultant anaesthesia lasting 25–45 min – but responses will be variable depending on species, solution concentration and temperature. One study in bronze frogs (*Rana clamitans*) and leopard frogs (*Rana pipiens*) used 1.5 mg/ml MS-222



Figure 17.9 • Induction of anaesthesia in a marine toad (*Bufo marinus*), in a plastic zip-lock bag using MS-222.

Table 17.4: Planes of anaesthesia

PLANE	REFLEXES RETAINED	REFLEXES LOST
Light	Withdrawal reflex, spontaneous movement, gular respiration, cardiac impulse	Righting reflex and corneal reflex
Deep	Cardiac impulse	Withdrawal reflex (last reflex to be lost), spontaneous movement, gular respiration, corneal reflex and righting reflex
Overdose	None	Cardiac impulse slow or difficult to detect

(Holmes and Pitham, 2004; Wright, 2006)

solution and produced surgical anaesthesia in 6–12 min (Letcher and Amsel, 1989). After removal from the solution, anaesthesia lasted 18–36 min and recovery was complete in 39–69 min.

Once the appropriate level of anaesthesia has been achieved, the animal should be removed from the induction solution or excessively prolonged anaesthesia may result. The patient's skin should be kept moist using well-oxygenated clean water. Further contact with anaesthetic agents should only be used if the animal shows signs of recovery before the procedure is completed. Artificial slime (for example Shieldex®, Aquatronic, Oxnard, CA) may be used to coat the skin. Water-soluble gels (for example KY Jelly®) may inhibit cutaneous respiration and so should not be used to coat the skin (Wright, 2001d).

A more dilute concentration of the anaesthetic may be syringed over the patient to prolong anaesthesia (Pizzi and Miller, 2005). Alternatively, isoflurane or halothane or

sevoflurane may be diluted (1:3 in water) and trickled over the patient; however, this option poses more health and safety issues for staff exposed to evaporating anaesthetic agents.

Rinsing the patient with clean water will reverse the anaesthetic and speed recovery. Recovery usually occurs in 30–90 min after removal of the anaesthetic (Hadfield and Whitaker, 2005). If necessary, doxapram hydrochloride can be administered to stimulate breathing (Raphael, 1993).

MS-222 has also been used intramuscularly or intracoelomically to induce anaesthesia. Doses required again vary between species, with 100–200 mg/kg being used in leopard frogs (*Rana pipiens*) and up to 400 mg/kg in bullfrogs (*Rana catesbeiana*) (Letcher, 1992). The solution was 125 mg/ml in sterile water, producing a pH of approximately 1.75. No peritonitis or inflammation was seen in the study. However, the duration of anaesthesia may be prolonged and cannot be 'reversed' by rinsing the patient as with induction using a solution.

An overdose of buffered MS-222 at 10 g/L in a 30-min bath appears to be appropriate for humane euthanasia of amphibian species (Hadfield and Whitaker, 2005).

Clove oil (eugenol)

A solution of clove oil has been used to induce anaesthesia in several species. Anecdotal reports suggest 318–400 mg/L for anaesthesia lasting 30–100 min in anurans and salamanders (Mitchell, 2003).

In leopard frogs (*Rana pipiens*) a bath concentration of 310–318 mg/L for 15 min induced surgical anaesthesia (Lafortune et al., 2001). The anaesthetic duration was extremely variable, from less than 5 min up to 65 min. A common side effect was gastric prolapse.

Benzocaine

In order to produce a solution for anaesthesia, benzocaine must first be dissolved in acetone. A stock solution can be prepared by adding 40 g benzocaine to a litre of acetone and protected from the light in a dark glass bottle; 10 ml of the stock solution mixed with 8 L of water will produce a solution containing 50 ppm benzocaine. This concentration will cause sedation in many species. Induction of anaesthesia may require a higher concentration, up to 200 ppm.

One study performed on cane toads (*Bufo marinus*) showed significant cardio-respiratory changes during benzocaine anaesthesia (Anderson and Wang, 2002). Apnoea occurred during anaesthesia after immersion in 1 g benzocaine/L for 15 min. PaO_2 dropped below 30 mmHg and respiratory acidosis developed. Acid–base status did not stabilise until 24 h after anaesthesia.

Ethanol

Immersion in 5–10% ethanol may produce anaesthesia. 80 proof vodka is 40% ethanol and can be diluted in fresh chlorine-free water to produce the appropriate concentration.

Volatile agents

Isoflurane, halothane and methoxyflurane

Volatile agents can be used to anaesthetise amphibians (Whitaker et al., 1999). Both isoflurane and sevoflurane depress myocardial conduction by affecting calcium channel currents (Hirota et al., 1996). There is a risk of damage to the patient's epidermis during anaesthesia with these agents, and self-trauma may occur with the irritation. There are also safety issues for staff with this form of anaesthesia unless the gases are provided via an endotracheal tube.

Usually the volatile agent is bubbled through water into which the animal is placed. Placing the animal on moist paper in an induction chamber will also induce anaesthesia. Care should be taken not to allow the patient to desiccate with the dry gases during this technique. Alternatively some of the anaesthetic liquid can be mixed with water-soluble gel before topical application to the amphibian. It is more difficult to titrate the dose with this technique and the gel must be wiped off to prevent further absorption once the patient is anaesthetised.

Injectable agents

Propofol

This agent may be administered by intracoelomic or intravenous injection, or applied topically. Propofol has been used to induce sedation or light anaesthesia in leopard frogs (*Rana pipiens*) at 10 mg/kg when injected perivascularly in the area of the sublingual plexus (Lafortune et al., 2001). Intracoelomic administration of propofol resulted in sedation (at 9.5 mg/kg) or anaesthesia (at 30 mg/kg) in White's tree frogs (*Pelodryas caerulea*) (von Esse and Wright, 1999).

Ketamine

Anaesthesia with ketamine is possible in amphibians and may be administered intramuscularly, intravenously, into the lymphatic system or subcutaneously. However, effective doses vary greatly both intra- and inter-specifically, and induction and recovery times are also variable (Whitaker et al., 1999).

Tiletamine/zolazepam

The combination of tiletamine with zolazepam produces anaesthesia in many species. When this was trialled in two amphibian species, bullfrog (*Rana catesbeiana*) and leopard frog (*Rana pipiens*), results were extremely variable (Letcher and Durante, 1995). There was profound intraspecies variation in depth and duration of effect. Doses that produced anaesthesia in some individuals were fatal in others. There are reports of using this combination at 5–20 mg/kg intramuscularly or subcutaneously to induce anaesthesia in anurans, but results are variable between species (Schumacher, 1996).

Medetomidine

In a study in leopard frogs (*Rana pipiens*) this agent failed to produce sedation or anaesthesia (Lafortune et al., 2001).

Cooling

Cooling amphibians will slow their metabolism and ease restraint, but does not provide any analgesia and may cause potential long-term immunosuppression (Green and Cohen, 1977). Cooling is not recommended as a form of anaesthesia.

Anaesthetic maintenance

If the heart rate or oxygen saturation decreases or metabolic variations are seen, oxygen flow to the patient should be increased and the anaesthesia reduced. Oxygen is provided by bubbling gas through the anaesthetic solution bathing the patient (cutaneous respiration), via an endotracheal tube (pulmonic respiration), or across the skin (cutaneous respiration). Anaesthesia is reduced by removing the animal from contact with the anaesthetic (as gas, solution, or topically), by changing to a solution containing a lower concentration of anaesthetic, and/or by flushing the animal with fresh water that does not contain anaesthetic.

Recovery

Recovery is complete when the patient has normal respiratory movements and is alert and responsive (Wright, 2001c).

Suggested anaesthetic protocols

Tricaine methanesulfonate is the anaesthetic agent of choice in amphibians. It can be used for induction and,

Table 17.5: Amphibian anaesthetic agents

DRUG	DOSE (concentration for bath or mg/kg)	ROUTE	COMMENT
Benzocaine	–	Bath	Dissolve in ethanol or acetone before water Use product from chemical suppliers or fish anaesthetic preparation; do not use topical mammal products
	50 mg/L[3]		Larvae
	200–500 mg/L[2,3]		Frogs, salamanders
Clove oil (eugenol)	0.3–0.45 mg/L[1,5]	Bath	Deep anaesthesia in leopard frogs and tiger salamanders
			Reversible gastric prolapse in 50% leopard frogs
Ethanol	5–10%	Bath	–
Halothane	4–5%[3]	Induction chamber	–
	Bubbled into water to effect[7]	Bath	
Isoflurane	3–5% (induction); 1–2% (maintenance)[6]	Chamber	Terrestrial species
	0.28 ml/10 ml bath[8]	Bath	Aquatic species
	3.0 ml + 3.5 ml KY jelly + 1.5 ml water[8]	Topical	Remove excess once anaesthetised
Ketamine	50–150 mg/kg[3]	SC, IM	Induction and recovery can be prolonged Variable response
Propofol	100–140 mg/kg[1]	Topical	Sedation to deep anaesthesia; rinse when desired plane achieved; recommended only for animals <50 g
	10–30 mg/kg[9]	I/Ce	White's tree frogs; sedation/light anaesthesia at lower dosage; recovery in 24 h

(*Continued*)

Table 17.5: (Continued)

DRUG	DOSE (concentration for bath or mg/kg)	ROUTE	COMMENT
Tricaine methanesulfonate (MS-222)	100–200 mg/L[10]	Bath to effect	Induction of larvae
	200–500 mg/L[3]		Induction of tadpoles, newts
	1 g/L[4]		Induction of most gill-less adults
	0.5–2.0 g/L[3]		Induction of frogs, salamanders (15–30 min)
	2–3 g/L[11]		Induction of toads (15–20 min)
	50–200 mg/kg[4]		May be irritant SC or IM[4]
Tiletamine/zolazepam	5–20 mg/kg[6]	IM, ICe	Variable results

Key: ICe = intracoelomic, IM = intramuscular, SC = subcutaneous
[1] (Carpenter, 2005); [2] (Cooper, 1984); [3] (Crawshaw, 1993); [4] (Downes, 1995); [5] (Lafortune et al., 2000); [6] (Schumacher, 1996); [7] (Stein, 1996); [8] (Stetter et al., 1996); [9] (von Esse and Wright, 1999); [10] (Whitaker et al., 1999); [11] (Wright and Whitaker, 2001)

at a lower concentration, for maintenance of anaesthesia. Depth of anaesthesia is easily changed by using differing concentrations of agent for immersion or cutaneous application. The nares should be kept above the level of solution to ensure inhalation does not occur.

ANAESTHESIA MONITORING

Observations on the patient

Cardio-respiratory systems

The minimal parameters that should be monitored during amphibian anaesthesia are heart rate, buccopharyngeal or pulmonic respiratory rate, and blood oxygen saturation (Wright, 2001c).

The *cardiac impulse* is often visible and heart rate can be easily recorded. Doppler flow machines are very useful in situations where the cardiac impulse cannot be observed, for example when the patient has been draped for surgery. A small 'footprint' (i.e. small patient contact surface) probe is required, as most amphibian patients are small.

Depending on the species, a *respiratory rate* may be obtained from observing buccopharyngeal or pulmonic movements. During anaesthesia, respiration should be slow and regular, but many anaesthetics will induce apnoea in amphibians. Hyperoxic conditions usually cause a cessation of respiratory movements, as hypoxia is often the drive for respiration. In order to stimulate respiration, room air should be used (rather than 100% oxygen) to stimulate respiratory movements during recovery (Wright, 2001c). Ideally, respiration may be slow during surgical anaesthesia, but remains regular despite painful stimuli. With some anaesthetics, spontaneous respiration will be lost; in these patients, intubation should be performed if possible to allow intermittent positive pressure ventilation (IPPV) (Crawshaw, 1993; Downes, 1995).

Central nervous system

Reflexes can be monitored in amphibians similarly as in other species (Lafortune et al., 2001). Loss of the righting reflex on induction denotes onset of anaesthesia (see Table 17.4). The corneal reflex is also lost at a light plane of anaesthesia. Surgical anaesthesia is deemed present when the righting reflex, withdrawal reflex and gular respiration are lost (Downes, 1995; Wright, 2006).

Anaesthetic monitoring equipment

Although not designed with amphibians in mind, many pieces of anaesthetic monitoring equipment can be used on these species.

With some *Doppler* flow detectors, heart sounds may be differentiated and characterised (Frye, 1994). Doppler ultrasound has been used to measure blood flow in the common iliac artery in bullfrogs (*Rana catesbeiana*) and cane toads (*Bufo marinus*) (Willens et al., 2006).

As most amphibian respiration is cutaneous, and pulmonic respiration often ceases under anaesthesia, it can be difficult to assess oxygen supply to the patient. During apnoea, oxygen uptake reduces, arterial oxygen falls, arterial carbon dioxide levels increase and arterial pH falls (Jones, 1972). If appropriate sized clips or probes are available, haemoglobin oxygen saturation may be measured using *pulse oximetry* devices. Small clips may be used on toes or tails, and probes may be placed on the ventrum over the heart or oesophageally. Amphibians tend to have lower oxygen saturation than mammals. No normal values exist, but Burggren and Just report levels over 95% in the tiger salamander (*Ambystoma tigrinum*) (Burggren and Just, 1992). As with other species, trends in values are important, and a decrease of 5% or more should prompt action to increase the patient's oxygen supply (Wright, 2001c).

Table 17.6: Analgesics that may be useful in amphibians

DRUG	DOSE (mg/kg)	ROUTE	DURATION	COMMENT
Buprenorphine	0.075[4] 38[2]	Dorsal lymph sac SC	>4 h	ED_{50} in leopard frog (*Rana pipiens*)
Butorphanol	0.2–0.4[3]	IM	–	–
Fentanyl	0.5[2]	SC	>4 h	ED_{50} in leopard frog (*Rana pipiens*)
Lidocaine (lignocaine) 1–2%	–	Local infiltration[1]		Local anaesthetic; use with caution
Morphine	30–100[5] 38–42[2]	IM, SC, topically SC	60–90 min >4 h	Little effect on feeding and behaviour
Nalorphine	122[2]	SC	>4 h	–

Key: IM = intramuscular; SC = subcutaneous

[1] (Johnson, 1991); [2] (Machin, 1999); [3] (Schumacher, 1996); [4] (Stephens and Rothe, 1997); [5] (Stevens et al., 1994)

Electrocardiogram (ECG) machines are not routinely used on amphibians, but may be used to monitor heart rate. ECG electrodes are positioned as in other species (Schoemaker and Zandvliet, 2005). Electrical conduction is enhanced by using needle electrodes. Reference values for anaesthetised animals have been published (Whitaker and Wright, 2001).

PERI-ANAESTHETIC SUPPORTIVE CARE

Patient management

An accurate weight (using digital scales) should be obtained prior to anaesthesia. Weighing should be repeated on a regular basis after recovery to monitor hydrational and nutritional status.

Most amphibians will use predominantly cutaneous respiration. Soak the amphibian in a shallow water bath for 60 min before anaesthesia to ensure adequate hydration (Wright, 2001d). The skin of amphibian patients should also be kept moist throughout the procedure, either with maintenance anaesthetic solution or high-quality water, to prevent desiccation and associated respiratory compromise. For species relying more on branchial respiration, for example the axolotl (*Ambystoma mexicanum*), the delicate external gills should be protected from trauma and desiccation.

Fasting

As the larynx is usually closed during anaesthesia, aspiration after regurgitation is very rare in amphibians. Nevertheless, loss of electrolytes via emesis should be avoided by pre-anaesthetic fasting if possible. Insectivorous species weighing less than 20 g should be fasted for no longer than 4 h, those weighing more than 20 g for 48 h, and those on a vertebrate diet for seven days (Wright, 2001c). Obviously these guidelines will vary depending on the patient's status, the anticipated period of anaesthesia and the procedure to be performed.

The environmental temperature will also affect metabolic rate and gastrointestinal transit time in these ectothermic animals. Amphibians maintained at temperatures below their preferred body temperature will require an extended fasting period – up to 10 days in larger specimens (Jiang and Claussen, 1993).

Analgesia

Amphibians display appropriate behaviour in response to a painful stimulus and possess antinociceptive mechanisms to modulate pain. These physiological findings suggest that pain perception in amphibians is likely similar to other vertebrates and analgesia should be provided for painful procedures (Stevens et al., 1994; Stoskopf, 1994). As with many other exotic pets, drug pharmacokinetics and doses have not been experimentally ascertained, but clinical investigations have proposed dose rates for various species.

The opioid ligand and receptor are conserved throughout vertebrate phylogeny, suggesting the potential benefits of opioids in amphibian analgesia (Stevens, 1988). Alpha-2-agonists, ketamine and ms-222 have also demonstrated analgesic potential (Brenner et al., 1994; Lee and Frank, 1991; Letcher, 1992).

Morphine, a μ-agonist, has been demonstrated to produce analgesia when administered intramuscularly, subcutaneously or topically (Stevens et al., 1994). However, variations in response between species are likely to exist.

EMERGENCY PROCEDURES/DRUGS

Respiratory problems

Gas exchange can be supported by increasing ambient oxygen levels or bubbling oxygen through water in the vivarium (Crawshaw, 1998). If apnoea is prolonged respiration can be stimulated using doxapram intramuscularly or intravenously at 5 mg/kg (Hadfield and Whitaker, 2005).

Cardiovascular problems

Bradycardia may be treated with atropine at 0.03 mg/kg intramuscularly (Hadfield and Whitaker, 2005). Adrenaline (epinephrine) can be administered intratracheally along with cardiac compressions if asystole occurs (Hadfield and Whitaker, 2005).

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18 Fish anaesthesia

Matthew Fiddes

INTRODUCTION

There are at least 24 600 known species of fish (Helfman et al., 1997), with over 2000 species entering the ornamental fish trade (Davenport, 2001). The total population of ornamental fish in the UK has been estimated at 135 million, with approximately 14% of UK households owning either an aquarium or pond (Anonymous, 2007). The bulk of this pet fish trade is in freshwater fish, split between coldwater species (such as koi carp [*Cyprinus carpio*] and goldfish [*Carassius auratus*]) and tropical species kept in aquaria. This is reflected in the fish species most often presented to veterinary surgeons. With continuing advances in the field of ornamental fish medicine, together with the large number of fish kept as pets and in public aquaria, there is great potential for further veterinary involvement.

Anaesthesia is a useful tool to the fish practitioner and enables various tasks to be performed. Fish often struggle in most forms of restraint and handling, which can result in damage to the fish and, in some cases, to the handler. Anaesthesia can greatly facilitate examination, transport and diagnostic sampling as well as reducing stress to the fish. Surgery will require anaesthesia and, with appropriate techniques, the fish may be maintained out of water for extended periods.

ANATOMY AND PHYSIOLOGY

As discussed, a large number of species are commonly kept in captivity and it is often this diversity that makes fish such attractive pets. Unlike many terrestrial environments, aquatic environments can be subject to a huge variation in conditions and this has resulted in a wide range of physiological differences. Water conditions can have a great effect on the homeostatic mechanisms in the fish.

Thankfully many anatomical and physiological features that are important in anaesthesia are common to most fish. Some clinically important variations will be discussed. The koi carp (*Cyprinus carpio*) is a species commonly presented for veterinary attention and is used to demonstrate typical external and internal gross anatomy (Fig. 18.1).

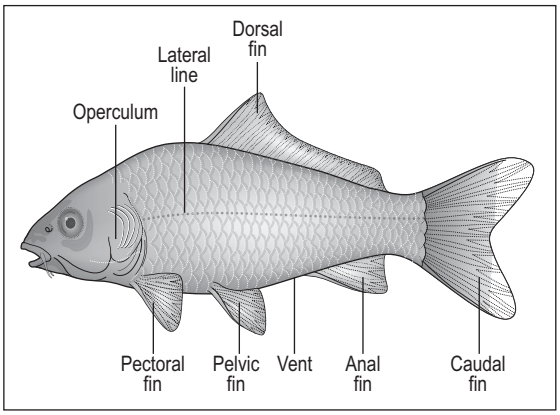


Figure 18.1 • The external and internal anatomy of the koi carp (*Cyprinus carpio*). (A) External anatomy. (B) Internal anatomy.

If the practitioner is unfamiliar with the species or variant presented it can often be useful to consult texts as to the normal anatomy of these fish. In particular, selective breeding of fancy goldfish has led to a wide variety of often extreme body types that may be mistaken for pathology (Fig. 18.2).

Temperature regulation

Fish, with the exception of some species, such as those in the suborder Scombroidei (which includes tuna), are ectothermic, so their body temperature varies with that of the surrounding environment. This has important implications for the speed of physiological processes. In the context of anaesthesia, drug metabolism may vary markedly with different temperatures; dosage intervals and absolute dosages may need to be altered accordingly. Meat withdrawal times for drugs used in farmed species for human consumption are expressed in degree days to reflect the varying speed of metabolism at different temperatures.

Cardiovascular system

Compared to mammalian species, the circulatory system is relatively simple in fish. Deoxygenated blood passes from the heart to the gills, where it is oxygenated by a counter-current system in the gill lamellae (Stoskopf, 1993a). From the gills, blood travels directly into the systemic circulation (Wildgoose, 2001a) and returns to the heart via the venous system.

The fish heart is often positioned quite cranially (Stoskopf, 1993a). It is a two-chambered organ with four distinct regions, and lies within the pericardial sac (Wildgoose, 2001a). Venous blood enters the thin-walled sinus venosus and subsequently the atrium. Blood then enters the thick-walled, muscular ventricle and is pumped into the bulbus arteriosus. The elastic recoil of the bulbus arteriosus pushes blood though the ventral aorta to the gills. There are a variety of vascular patterns in fish and most have well developed renal or hepatic portal systems (Wildgoose, 2001a). The renal portal system may have effects on the pharmacokinetics of drugs administered intramuscularly or intravenously at various sites.



Figure 18.2 • The normal appearance of an Oranda, a variety of goldfish (*Carassius auratus*). Selective breeding has resulted in wide anatomical variation of the same species.

Respiratory system

With few exceptions, the major respiratory organs are the gills (Childs and Whitaker, 2001). These are the site of exchange of respiratory gases, but are also vital for nitrogenous waste excretion, acid–base balance and osmotic control. The gills consist of multiple bony/cartilaginous gill arches (Fig. 18.3) and typically lie either side of the pharynx in the pathway of water from the mouth to the exterior of the animal. The cranial aspects of the gill arches are modified into gill rakers that allow separation of food particles from the water as it flows over the gills (Stoskopf, 1993a). The

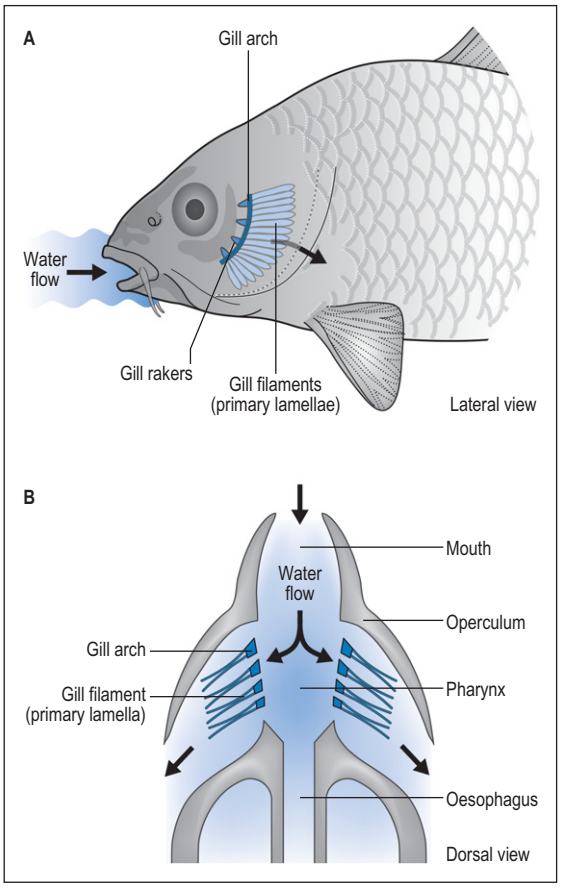


Figure 18.3 • Anatomy and water flow over the teleost (bony fish) gill. Water passes from the buccal cavity over the gills and exits via the opercular clefts. Blood travels against the water flow at the secondary gill lamellae to provide an efficient counter-current mechanism of respiratory gas exchange. (A and B) Water flow over the gills.

(Continued)

gill filaments (primary lamellae) project caudally from the gill arches, giving the gills a comb-like appearance. Each gill filament has secondary lamellae lying perpendicular to its axis, which act as the site of diffusional/osmotic exchange. A secondary lamella consists of two layers of epithelial cells surrounding a vascular space. The arrangement of the secondary lamellae and the associated blood supply acts as an efficient counter-current mechanism of gaseous exchange. The gills are often protected from the exterior by bony plates (operculae).

Oxygen concentration in water is typically much lower than the oxygen concentration in air experienced by terrestrial animals. The gill arrangement, with its counter-current mechanism, allows extremely efficient removal of oxygen (Harms, 2003). This efficiency can, however, damage the fish if dissolved oxygen levels are very high (supersaturation) (Mayer, 2005). In a clinical context, care should be employed in attempting to oxygenate the water. It is preferable to bubble air through the water via an air-stone or diffuser rather than using 100% oxygen.

Water must be passed over the gills to maintain their respiratory and excretory functions. Mostly water is drawn in through the mouth, passes over the gills and exits caudally through the gill openings (opercular clefts). This is a unidirectional flow of water (Childs and Whitaker, 2001) and directing water in the opposite direction may damage the sensitive gills. This is important to consider in ventilation/resuscitation where the fish should not be dragged backwards through the water (Brown, 1988) and water should only be directed by pump or syringe though the mouth and not through the gill openings. Water flow across the gills is chiefly achieved by the opening and closing of the mouth and opercula, together with movements of the pharyngeal floor. Movement of the opercula is often the best guide to ventilatory effort and is referred to as the respiratory rate. In some species movement of the mouth, in particular with the presence of pharyngeal valves, is crucial in allowing water to pass over the gills (Stoskopf, 1993a). This may be of relevance to artificial ventilation when movement of the mouth may be required to facilitate water passage over the gills (Harms, 2003).

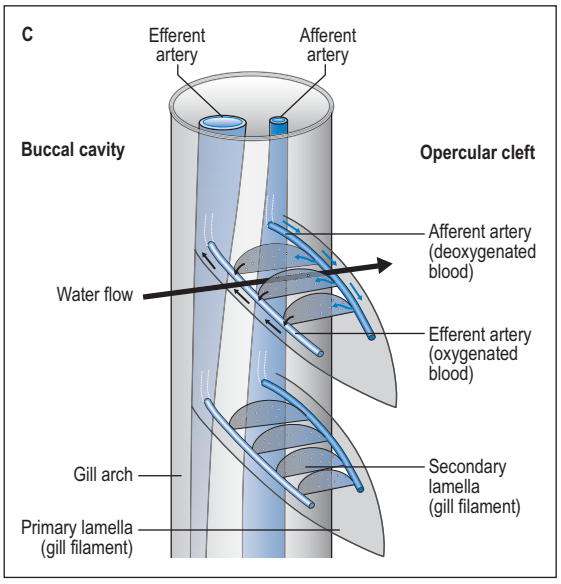


Figure 18.3 • (Continued) (C) Water flow and blood flow in the gill lamellae.

The gills do not function well out of water. Gill lamellar collapse occurs out of water, which decreases the exchange area for absorption of gases across the lamellar membrane (Brown, 1993). If the fish is removed from the water for more than a few minutes, steps must be taken to pass water over the gills to allow respiratory/excretory exchange and to prevent desiccation.

Some fish are capable of utilising atmospheric oxygen (Childs and Whitaker, 2001) instead of dissolved oxygen in the water and may be referred to as 'air-breathers'. This can make induction with waterborne anaesthetic agents problematic (for example, snakeheads, family Channidae) (Ross, 2001).

Osmotic homeostasis and electrolyte balance

The major organs involved in osmotic regulation are the gills, kidneys and skin. In most species the skin provides a relatively impermeable barrier, reducing the area of osmotic exchange. The gills and kidneys have varying roles dependent on surrounding water conditions.

Fish are generally classified into freshwater or marine species, but some are capable of switching between these environments and some inhabit estuarine/brackish conditions. The degree of salinity has a major role in determining the strategy of maintaining electrolyte balance in fish.

Freshwater fish are effectively immersed in a hypotonic solution and, therefore, are faced with water influx by osmosis across exposed epithelial surfaces, in particular the gills. Ion transport across the gills is utilised to maintain osmotic regulation (Childs and Whitaker, 2001) and the kidneys excrete urine that is primarily water, thus eliminating excess water (Mayer, 2005). Extensive disruption of osmoregulation, such as damage to environmental barriers or to the organs responsible for osmotic control, can potentially result in ascites, with or without cutaneous oedema (Wildgoose, 2001a). This may be observed as the clinical condition referred to as 'dropsy' where there is coelomic (abdominal) distension and raised scales, giving a 'pine cone' appearance. One possible aetiology is an internal bacterial infection.

Marine fish are immersed in a hypertonic solution and face the reverse problem to freshwater fish (Fig. 18.4). There is osmotic loss of water mainly from the gills, together with diffusional entry of salt. This is countered by the fish drinking seawater to replace the lost water. However, this is inevitably accompanied by a large intake of salt. Specialised chloride cells found in high concentrations at the gills (Childs and Whitaker, 2001) actively transport sodium and chloride ions out of the fish. The major role of the kidney is excretion of excess electrolytes, mainly magnesium and sulphate (Mayer,

2005). If there are skin wounds or major gill damage in marine fish, there may be a rapid loss of water,and so clinical management may need to involve fluid replacement.

Integumentary system

Most fish possess scales, which are flexible bony plates that develop in scale pockets in the dermis (Wildgoose, 2001b). Large scales on fish, such as sturgeon, can make injections difficult. Some species are 'scaleless' and often have thicker epithelial skin layers to compensate. Such species may be more sensitive to waterborne drugs and toxins. The epidermis and cuticle (outermost skin layer) form the waterproof barrier. As discussed previously, damage to the skin breaches this barrier and may disrupt osmotic control.

Epithelial cells secrete mucus to form the cuticle, which also contains antibodies and lysozymes that have antibacterial and antifungal properties (Wildgoose, 2001b). This skin coating can easily be damaged by rough handling, leaving the fish more vulnerable to pathogen invasion.

Pain

The subject of whether fish can feel pain has been a continuing subject of debate. However, there have been studies to suggest that fish are capable of experiencing pain (Chandroo et al., 2004; Dunlop et al., 2006; Sneddon, 2003a). The presence of nociceptors with similar physiological properties to those found in higher vertebrates has been demonstrated in fish (Sneddon, 2003b). As in varying mammalian species, the expression of pain may well be varied in fish and not amenable to anthropomorphic comparison. Pain responses attributed to application of noxious chemicals to the lips of trout (*Oncorhynchus mykiss*) included anomalous behaviours where they rocked on either pectoral fin from side to side and also rubbed their lips (Sneddon, 2003a). Opercular beat rate (respiratory rate) was also increased. Administration of morphine appeared to reduce these pain responses and this was attributed to the analgesic properties of morphine.

This is likely to be an area of continuing debate and research, but it would seem justifiable to take steps to minimise pain and suffering in fish patients. Also important to consider in a pet animal context is the owner's perception of the veterinary care provided, which often factors analgesia as beneficial.

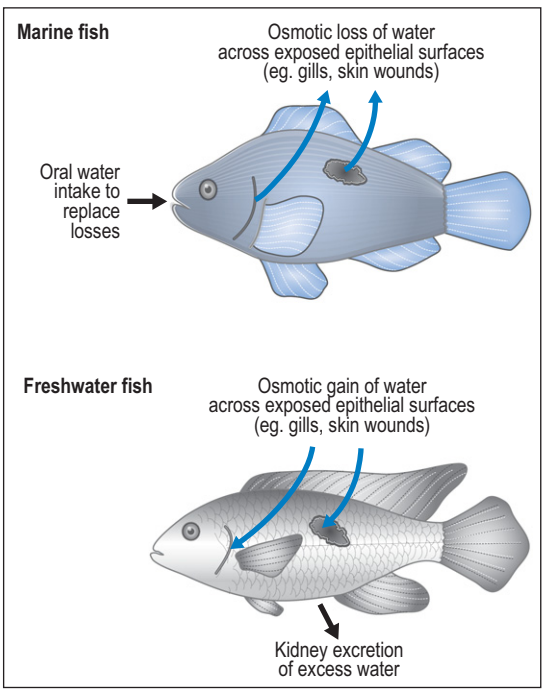


Figure 18.4 • Demonstration of water flow in osmoregulation by freshwater and marine fish. Freshwater fish are effectively immersed in a hypotonic solution and are subject to osmotic water influx. Marine fish face the reverse situation as they are immersed in a hypertonic solution.

Health and safety

Some species of fish have potential hazards to personnel involved in handling. Injury may be caused by venomous spines (for example, the lionfish [*Pterois volitans*]), sharp spines, teeth and electrical charges (for example, the electric eel [*Electrophorus electricus*]) (Grist, 2001). Knowledge of the species involved and an appropriate risk assessment should make anaesthesia of these fish as safe as possible. Hazardous species should only be handled with more than one person present, and all team members aware of specific first aid measures.

Possible zoonoses should also be considered, such as *Mycobacterium* spp. that may cause cutaneous infections in humans (Grist, 2001). In general, good handling practice, in particular the wearing of gloves, should minimise the risk.

BOX 18.1 Anatomy and physiology

- Gills are the major respiratory organ
- Gills are also involved in nitrogenous waste excretion, acid–base balance and osmotic control
- There is unidirectional flow of water from the mouth, over the gills and exiting through the opecular clefts
- Opercular (gill cover) movements are referred to as the respiratory rate
- Freshwater fish are immersed in a hypotonic solution and marine fish in a hypertonic solution. Homeostatic mechanisms are required to maintain osmotic balance
- Skin is mostly impermeable and provides an osmotic barrier, as well as a barrier against pathogen invasion
- Most fish are ectothermic
- The circulatory system is relatively simple, often with renal portal system

PRE-ANAESTHETIC ASSESSMENT AND STABILISATION

History and clinical examination

The owner should be questioned regarding current and prior husbandry conditions, and any previous medical problems. This information will allow the clinician to identify any factors that may predispose disease.

In contrast to many domesticated species, fish will be stressed by handling. This means that a 'hands-on' approach to pre-anaesthetic checks is unlikely to be rewarding. However, much information can be observed by remote assessment. If possible, fish should be observed in their normal surroundings and an assessment made in comparison to the other fish. If all fish are showing a clinical sign then this may indicate possible adverse water quality. Assessment of water parameters is often performed as part of a fish health investigation and may be useful prior to anaesthesia. In particular ammonia, nitrite, pH and temperature should be measured (Cecil, 2001). It should be remembered that the water in which a fish has been transported is likely to have been fouled and so will not be representative of the conditions in the pond or tank of origin.

As will be discussed, fish anaesthesia often involves waterborne anaesthetic agents that rely on uptake and excretion by the gills. Therefore, compromised gill function may have an effect on anaesthesia, as well as compromise of respiratory and homeostatic functions. Signs of gill disease (although often non-specific) include increased respiratory rate (increased opercular beating), gaping at the surface and congregation at waterfall/filter inlets. The gills may be visually inspected for signs of damage (Fig. 18.5). Diagnostic procedures and handling should be minimised until the critically hypoxic patient is stabilised (Childs and Whitaker, 2001).

Fish cases are often presented with advanced systemic disease and pathology is frequently present in the absence of any obvious clinical signs (Wildgoose, 2001a). This should be considered in pre-anaesthetic assessment.



Figure 18.5 • Normal appearance of healthy gills in a goldfish (*Carassius auratus*). The operculum (gill cover) has been lifted. Note the uniform red colour of the gill filaments.

Many fish will be transported to veterinary surgeries for initial presentation. Proper handling and transport at this stage, and subsequently, will help minimise stress and prevent iatrogenic damage to the fish.

Fish should be handled with latex gloves to minimise damage to the skin or mucus covering. Fine mesh nets large enough to support the fish should be used and sock nets may be employed for larger fish. Covering the head and eyes improves handling. Catching, transport and waiting time should be minimised to reduce stress to the fish. Transport containers should be large enough to accommodate the fish comfortably – tied polythene bags placed in polystyrene boxes are commonly used. For short journeys the bag may be nearly completely filled with water; however, for longer journeys a ratio of ⅓ water to ⅔ air (or oxygen if available) is preferable to help maintain water oxygen concentrations. Fish may be starved before transit to prevent fouling of the water, with food being withheld for one feeding cycle prior to transport or anaesthesia (Brown, 1988). For veterinary surgeons in general practice, appointments for fish clients should ideally be made outside normal consultation times to allow anaesthetic procedures to be carried out, where appropriate, as soon as possible after initial assessment.

Weight measurement can be problematic and is often best performed following sedation or anaesthetic induction where the fish can be easily removed from the water and placed on a balance. Length-to-weight charts are available for some species and can be useful in estimating approximate weights (Table 18.1).

It is recommended that written informed consent for anaesthesia is obtained from the owner.

BOX 18.2 Pre-anaesthetic assessment

- Good history, including medical and husbandry conditions, is important
- Examine fish, usually by remote assessment, as they are stressed by handing
- Fish are often presented with advanced systemic disease
- Water quality assessment is useful, including temperature, pH, ammonia, nitrite and dissolved oxygen
- Good handling and transport minimise stress and iatrogenic damage

Table 18.1: Guide to weight of koi (*Cyprinus carpio*), from length of fish. These are only approximate guidelines and considerable variation will occur. The use of tables such as this is not a substitute for accurate weighing of fish

SIZE (cm)	30	35	40	50	55	60	70
BODY WEIGHT (kg) – MALE	0.6	1	1.5	2	3	4	5
BODY WEIGHT (kg) – FEMALE	0.8	1.25	1.9	2.5–3	4.5	5.5–6	7

(Holmes and Pitham, 2004)

Hospitalisation

Hospital tanks at the veterinary surgery may be used to house fish before and after anaesthesia, with the advantage of more intensive treatment and observation by trained medical staff (Stoskopf, 1993c). The water quality should be well maintained in such a setup with adequate filtration and heating as appropriate. Tanks will need to be sterilised between patients, which will inevitably kill established biological filters. For this reason biological filters are, in general, not useful. These problems, together with the stress of changing the fish's environment and water conditions, mean that the patient is often best returned to the tank or pond of origin when recovery is complete. Often owners will have hospital enclosures of their own that, providing water quality can be maintained, are useful to house recovering fish isolated from the main stock. Provision should be met for fish that are not content to be solitary.

Fluids

Parenteral fluid therapy is largely contraindicated in freshwater fish as there is already a net osmotic gradient into the body from the surrounding water. Adding a small amount of salt to the water in which the fish is immersed may help to reduce this osmotic gradient and reduce net water gain in fish that have damage to osmotic barriers or osmotic homeostatic sites (such as the gills, skin or kidneys) (Lewbart, 2001c; Lloyd, 2001). Fluid therapy may, however, be appropriate in saltwater species where the hypertonic environment can make the animal subject to fluid loss, particularly with gill and skin damage (Table 18.2).

The use of parenteral fluids has been described in dehydrated sharks either by intravenous or intraperitoneal/intracoelomic administration using 5% dextrose solution (Stoskopf, 1993d). Fluid rates should be balanced to the degree of dehydration (seen in sharks as elevated haematocrits and/or serum urea nitrogen levels), but 20 ml/kg/day is a safe starting point (Stoskopf, 1993d). Blood urea nitrogen produced by the liver is important in shark osmoregulation and levels may fall in cases of hepatic dysfunction. Elasmobranch balanced salt solution, an electrolyte solution with physiological concentrations of sodium chloride and urea added, has been used in transported sharks (Andrews and Jones, 1990), and this solution may be appropriate in situations of hepatic dysfunction or where electrolyte replacement is required.

BOX 18.3 Supportive care

- Hospital tanks are useful provided stress can be minimised and water quality maintained
- With freshwater species, addition of low doses of salt to the water may help reduce osmotic stress
- Parenteral fluid therapy is contraindicated in freshwater species as there is already an osmotic gradient into the fish
- Fluid therapy may be useful in marine species, especially if there is damage to natural osmotic barriers
- Studies suggest fish are capable of experiencing pain and so analgesic use is appropriate in potentially painful procedures

EQUIPMENT REQUIRED

The equipment required will vary with the anaesthetic modality used, with little specialist equipment required

Table 18.2: Fluids and supportive care in fish

DRUG	DOSE	COMMENTS
Fluid therapy		
Dextrose (5%)	20–30 ml/kg/day IV, IP[3]	Sharks; dehydration[3]
	40–60 ml/kg/day IV, IP[3]	Severe dehydration[3]
Lactated Ringer's	20–60 ml/kg/day IV, IP[3]	Sharks; dehydration; administer in divided doses[3]
Ringer's	18 ml/kg prn IV, IP[3]	Dehydration[3]
Dextran	20 ml/kg IV[3]	Hypovolaemia: given to effect[3]
Elasmobranch balanced salt solution (phenol red-free Hank's balanced salt solution (BioFluids, Rockville, Maryland) with 8 g/L NaCl and 31.02 g/L urea added)[1]	2 ml/min IP[1]	Mature sandbar sharks (*Carcharhinus plumbeus*)[1]
Miscellaneous agents		
Salt (sodium chloride)	1–3 g/L tank water[2]	Supportive measure in freshwater fish Use non-iodinised salt (not table salt)

Key: IM = intramuscular, IP = intraperitoneal (intracoelomic), IV = intravenous, prn = 'as the situation arises'

[1] (Andrews and Jones, 1990) [2] (Lewbart, 2001c) [3] (Stoskopf, 1993b)

for basic procedures. However, many waterborne anaesthetics are used exclusively for fish and will need to be obtained specifically for this purpose. Nets for handling and air pumps may be obtained from pet stores. Appropriate containers for induction or recovery chambers may be fashioned from any non-toxic watertight containers, with plastic storage boxes available from DIY stores working well for fish such as larger koi.

Transport and handling

Fish within a pond or tank are usually caught using a net. The clinician should wear latex or nitrile gloves when handling fish, to prevent damage to the sensitive cuticle. Two watertight containers, preferably with covers, should be available for use as induction and recovery chambers. The client should be asked to bring sufficient water from the fish's pond or aquarium to fill both the induction and recovery chambers. An air pump with air stone attached is used to aerate the water in the chambers.

Induction

Measuring spoons are useful to measure anaesthetic powders; those designed for culinary purposes work well. Accurate balances are also appropriate for this. Small syringes are used to measure liquids.

Maintenance

If the fish is to be maintained under anaesthesia out of the water, large syringes (for example, 60 ml catheter-tip syringes) can be used to direct water over the gills (Fig. 18.6). Non-abrasive surfaces, such as a waterproof drape or reversed incontinence pad, will prevent damage to the delicate piscine cuticle. A foam block with a V-shape cut out can be used to support the anaesthetised fish out of water.

For longer procedures, an anaesthetic delivery system may be used to direct water continuously over the gills. This may be constructed from an opened intravenous fluid bag with giving set attached, or a submersible aquarium pump with tubing attached.

Monitoring

Water quality may be measured before induction and during the procedure. A thermometer with an appropriate range for aquarium use will allow temperature to be assessed. Various test kits and meters are available to measure pH, oxygen, ammonia, nitrite and nitrate; these are useful, but not essential for individual fish anaesthetics.

An 8 MHz Doppler ultrasound probe is the most useful piece of equipment for monitoring the fish during anaesthesia (Fig. 18.7). An electrocardiogram (ECG) may also be used.

Resuscitation

A modified rubber enema pump may be used to deliver unidirectional water flow across the gills for resuscitation procedures.

TECHNIQUES

Injection sites

Skin preparation with agents such as alcohol and chlorhexidine prior to injection may cause irritation and skin damage, particularly in elasmobranchs (sharks and rays), as well as being ineffective (Stoskopf, 1993d). Irrigation with sterile

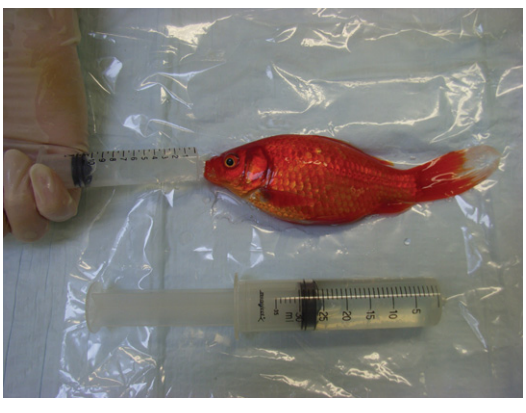


Figure 18.6 • A syringe containing anaesthetic solution can be used to maintain anaesthesia. The solution is applied into the mouth, so that water passes over the gills and out of the opercular clefts. This is the normal unidirectional flow of water – reversal of water direction reduces the efficiency of gas exchange and may damage the gills.

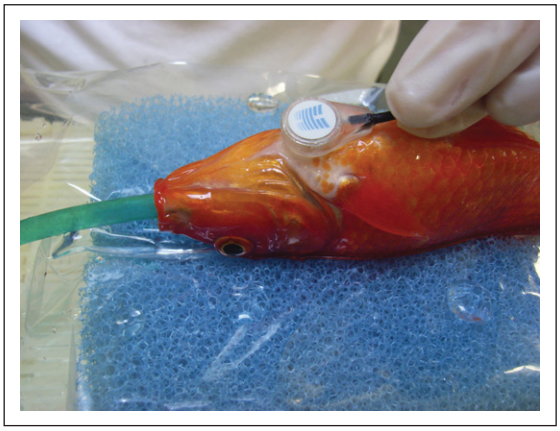


Figure 18.7 • Doppler probe positioned to monitor heart rate in a goldfish (*Carassius auratus*). Here the probe is positioned on the ventral midline between the pectoral fins.

saline may be performed prior to injection if there is gross contamination. Contamination of the needle before injection should be avoided.

Subcutaneous injections are generally not recommended in fish. The inelasticity of the skin reduces the potential space between the skin and muscle layers, and means the injected drug may be expressed through the injection site.

Intramuscular injections are best given into the epaxial muscles (Fig. 18.8). A position on the flank dorsal to the lateral line and lateral to the dorsal fin should be chosen. The needle should be aimed cranially, angled at 45°, and positioned between scales to reduce the chance of scale damage (Wildgoose and Lewbart, 2001). A dorsal midline approach is sometimes used where the needle is positioned cranially or caudally to the dorsal fin and injection made between the muscle fillets. Owners, particularly of valuable show koi carp (*Cyprinus carpio*), should be advised of possible scale loss caused by injection at these highly visible sites. Long needles should be used in large fish to minimise the risk of the drug being expressed through the injection hole. Intramuscular injection under the pectoral fin from a caudal approach is described in koi (Holmes and Pitham, 2004). This has the advantage of being on the ventral aspect of the fish, making any scale loss or scarring less visible. However, this is only suitable for small injection volumes and the proximity to vital internal structures should be remembered.

Intravenous access in fish may be problematic and is largely restricted to adequately restrained patients. The caudal vein lying ventral to the tail vertebrae may be accessed by a ventral midline or lateral approach. The veins on the medial aspect of the operculae of larger koi may be used for administration of euthanasia agents (Ross, 2001). The brachial arch may be a useful site for phlebotomy in teleost fish (Suedmeyer, 2006).

Intracoelomic injections are administered on the ventral midline between the vent and pelvic fins with the needle directed in a craniodorsal direction. A short needle (6–12 mm) is especially important in small ornamental species because of the proximity of the spleen to the point of injection (Brown, 1993). This route may be hazardous in koi and other carp because of the normal presence of peritoneal adhesions, which can result in injection into a solid organ or gastrointestinal tract (Wildgoose and Lewbart, 2001).

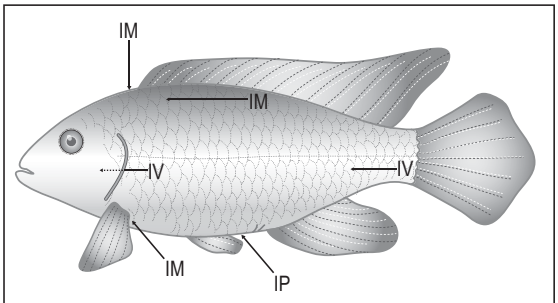


Figure 18.8 • Injection sites in fish (see descriptions in text).

ANAESTHETICS

Waterborne anaesthesia (see Table 18.6) is the most widely used method of fish anaesthesia and is analogous to gaseous anaesthesia in mammals (Ross, 2001). The anaesthetic agent in aqueous solution is ventilated by the fish and passes into the bloodstream across the gills. The agent is delivered to the central nervous system to produce anaesthesia. Most agents are excreted predominantly by the gills (Ross, 2001), but the liver, kidneys and skin may play roles in metabolism and excretion. In general, waterborne agents are more easily given to effect. The anaesthetic depth may be easily changed by using solutions containing different concentrations of anaesthetic agent.

Injectable agents (see Table 18.6) are most useful in larger fish, where waterborne agent induction may be impractical as large quantities of drug would be required to reach effective concentrations in large water systems (Stoskopf, 1993d). For longer procedures, waterborne agents may be used to maintain anaesthesia following injectable agent induction. Most injectable anaesthetics are associated with longer recovery times than waterborne agents. It is often difficult to gauge fish weights accurately prior to anaesthesia and, therefore, difficult to dose injectable agents accurately. Use of estimated doses of injectable anaesthetics may lead to over- or under-dosing, which may be of critical importance in smaller species.

If the fish is removed from water for more than a brief period, the same attention to maintaining water flow over the gills to enable respiratory gas exchange is required, independent of the mode of anaesthesia used.

Waterborne anaesthetic agents

MS-222 (ethyl-m-aminobenzoate, tricaine methanesulfonate)

MS-222 is the most widely used fish anaesthetic (Harms, 1999). It is licensed for fish in the UK and is approved for food fish use in the US (Harms, 2003). MS-222 is a white crystalline powder that is readily soluble, making anaesthetic solution preparation easier. MS-222 may be made up into a stock solution of 10 g/L, from which the required volumes can be used. The solution is unstable in light and so should be kept in a dark, stoppered container (Harms, 2003). In correct conditions this stock solution may be kept for up to 3 months (Ross and Ross, 1999). Refrigeration or freezing lengthens shelf life (Harms, 1999). Darkening of the solution (Ross, 2001) or the presence of oily residues indicates degradation.

If an accurate balance is unavailable or the stock solution is going to be used infrequently, then it may be more practical to use a standard pharmaceutical scoop (or, if necessary, a measuring spoon used for culinary purposes) to measure out the required amount of MS-222. A level 0.5 ml scoop holds approximately 400 mg of MS-222, and a

level quarter-teaspoon measure (equivalent to 0.8 ml scoop) will hold approximately 650 mg of MS-222 (Ross, 2001).

MS-222 solutions are acidic and so should be buffered before use with fish (Harms, 2003). Unbuffered solutions may cause hyperactivity and stress on induction (Ross and Ross, 1999). Saturation with sodium bicarbonate will buffer the solution to between pH 6.0 and 7.5 (Harms, 2003). Practically, a buffered stock solution may be prepared by adding sodium bicarbonate equal in mass to the amount of MS-222 used (Lewbart, 2001b). *Tris*-buffer may also be used.

Induction is rapid (approximately 1–2 min [Hall and Clarke, 1991], dependent on the concentration used) and MS-222 is regarded as having a good safety margin (Ross, 2001). Safety margins are reduced in warm, soft water (Harms, 2003), and also with young fish (Harms, 2003) and small fish (Ross and Ross, 1999). Recovery times are usually rapid, and equilibrium and motor activity can be expected to return after a few minutes. Prolonged recovery of up to 6 h may be expected after longer procedures. Also, as the drug is fat soluble, older or gravid animals may show slower recovery times. Anaesthesia induced with MS-222 may contribute to hypoxaemia, hypercapnia, respiratory acidosis and hyperglycaemia (Sladky et al., 2001).

MS-222 is absorbed across the gill epithelium and is biotransformed in the liver and probably the kidney (Harms, 1999). It is cleared primarily across the gills as the free and acetylated form, with additional metabolites being eliminated in the urine or bile (Harms, 1999). Tissue levels decline to almost zero in 24 h (Ross and Ross, 1999). However, withdrawal times should be observed if used with fish for human consumption.

MS-222 has been administered in sharks as a solution sprayed on to the gills with the fish in the water (Brown, 1993). However, prior restraint is required and is not appropriate for use in closed systems where the water may be heavily contaminated (Stoskopf, 1993d).

Benzocaine

Benzocaine is a white crystalline material that is similar to MS-222 (Ross and Ross, 1999). Unlike MS-222, it is almost totally insoluble in water and so must be dissolved in acetone or ethanol first (Ross, 2001). It is stable in a stock solution (usually 100 g/L) and can be stored in a sealed dark bottle for up to 1 year (Ross, 2001).

In solution benzocaine is neutral (Ross and Ross, 1999) and so usually does not require buffering. Benzocaine is almost identical to buffered MS-222 both chemically and in terms of physiological reactions.

Like MS-222, benzocaine has a good margin of safety, but this margin appears reduced at higher temperatures (Ross and Ross, 1999). Its efficacy is unaffected by water hardness or pH (Ross, 2001). Again benzocaine is fat soluble and recovery times can be prolonged in older or gravid animals.

Eugenol (clove oil)

This product is readily available without prescription (Ross, 2001). Over-the-counter pharmacy preparations contain approximately 90–95% eugenol (Harms, 2003). For calculation simplicity, it may be assumed that 1 ml clove oil contains approximately 1000 mg of eugenol (Lewbart, 2005). Eugenol is poorly soluble in water and so needs to be dissolved in ethanol before use (Lewbart, 2001b). A stock solution of 100 mg/ml may be achieved by diluting 1 part clove oil with 9 parts 95% ethanol. Eugenol may be used in freshwater and marine species.

Eugenol results in anaesthesia comparable to MS-222, but eugenol is characterized by more rapid induction, prolonged recovery and a narrow margin of safety (Sladky et al., 2001). The analgesic properties of eugenol are unknown. Fish may exhibit a calmer induction than with other agents, including MS-222 and benzocaine (Munday and Wilson, 1997). The use of high doses of eugenol (>100 mg/L) for induction of anaesthesia may be associated with rapid ventilatory failure, so care should be exercised. Commercially available, Aqui-S® (Aqui-S New Zealand Ltd, *www.aqui-s.com*) is a compound mixture of eugenol and polysorbate 80 (added for solubility) (Lewbart, 2005). This product can be added directly to water, usually having been prepared in a 10-fold diluted stock solution.

Owing to the wide availability of clove oil, owners may use it for euthanasia of fish. A dose of at least 10 drops per litre is recommended (Ross, 2001). The fish should be left in the solution for 10 min after respiratory (opercular) movements have stopped. It is suggested that prolonged immersion in high-dose clove oil solution is a more appropriate means of euthanasia by owners than carbon dioxide (see below). The owner should be advised of the insolubility of clove oil, and it may be mixed thoroughly with warm water to improve dispersion.

Carbon dioxide

Carbon dioxide (CO_2) is readily soluble in water and has been used as a sedative and anaesthetic in fish species. Carbon dioxide gas may be bubbled through water or compounds releasing CO_2 may be added to the water (sodium bicarbonate, Alka-Seltzer®, Bayer, Germany) (Harms, 1999). It can be difficult to control the dissolved concentrations. For controlled anaesthesia, dissolved oxygen has to be maintained at a high concentration, which has its impracticalities (Ross and Ross, 1999). Carbon dioxide also has adverse physiological effects compared to other anaesthetic agents, such as marked alteration of blood gases, acid–base balance (Iwama et al., 1988) and ECG (Mitsuda et al., 1988). For these reasons the use of carbon dioxide is not to be recommended for clinical anaesthesia. Baking soda and Alka-Seltzer® are common household products and may be useful as a last resort for anaesthesia. Again, fish should be left in the solution for 10 min after respiratory movements have stopped.

Fluorinated hydrocarbons

These compounds include isoflurane and halothane, and have the advantage of being readily available in most veterinary practices. They may be used by direct addition of the liquid to water (best achieved by spraying the liquid through a hypodermic needle under the surface) or by bubbling the vaporised compound through the water.

Halothane has been shown to provide rapid induction providing good surgical anaesthesia with excellent maintenance and fast recovery (2–5 min) (Ross and Ross, 1999). Despite these properties, the use of these agents has a number of disadvantages. Due to their relative insolubility anaesthetic levels are difficult to control. If added as liquids to the water they may remain in pockets in the water. This gives rise to the possibility of the fish ventilating potentially lethal concentrations of the agent (Brown, 1993). It is difficult to scavenge these gases in this application and so there is a potential hazard to operators.

Again the use of these agents should be a last resort, but may be potentially useful for restraint prior to the use of injectable euthanasia agents or percussion in situations where no other alternatives are available.

Injectable anaesthetic agents

Ketamine

Ketamine may be used as a single agent given by intramuscular injection. High doses are required in many teleost (bony fish) species and even then incomplete anaesthesia may result. This makes ketamine alone useful for restraint, but not as a replacement to waterborne anaesthetics for surgical procedures (Harms, 1999). Ketamine used in cichlids provided anaesthesia for up to 40 min; recovery required up to 4 h (Ross and Ross, 1999). The safety margin is reduced at higher temperatures. Respiratory support may be required with this agent.

Ketamine and alpha-2-adrenergic receptor agonist combinations

This combination reduces the total dose of ketamine required and reduces muscle spasms that can occur with ketamine alone (Stoskopf, 1993d). The use of medetomidine has the advantage of reversibility with atipamezole (Lewbart 2005). Ketamine combinations can cause mild bradycardia and apparent respiratory depression (Fleming et al., 2003). The use of alpha-2-agonists as sole agents is not recommended – studies with xylazine used at doses required for anaesthesia in trout showed ventilatory collapse, convulsions and gross ECG disturbance (Oswald, 1978).

Propofol

This is a widely used drug in small animal practice, but as in other species is administered by intravenous injection, which limits its use in fish species. Induction may be faster than ketamine combinations, with one study showing induction to a light plane of anaesthesia in 5 min (Fleming et al., 2003). Propofol showed more profound bradycardic and respiratory depression effects than ketamine combinations (Fleming *et al.*, 2003).

Alfaxolone–alphadalone

This drug has a number of advantageous properties. There is a stimulatory effect on the heart, providing a very regular and forceful heart beat (Ross and Ross, 1999). Vasodilatory properties are most obvious at the gills and aid in adequate oxygenation of the blood. At low doses (12 mg/kg) respiration and circulation were maintained at basal levels in certain species (Tytler and Hawkins, 1981). As with the ketamine combinations, the duration of anaesthesia is long (Ross, 2001). Again, intravenous administration often limits the use of this drug in clinical situations.

Lidocaine (lignocaine)

Lidocaine may be used as a waterborne anaesthetic, but there can be wide variation in dosages required between species (Carrasco et al., 1984). Its use as a waterborne agent is, therefore, not recommended, but it may be used as an injectable local anaesthetic, either alone or with sedative agents (Harms, 1999). Care must be taken not to overdose small fish when used by local injection (Harms, 2003).

BOX 18.4 Anaesthetics

- Waterborne anaesthetic agents are most widely used – analogous to inhalational anaesthesia in terrestrial animals
- The most commonly used waterborne anaesthetic is MS-222. It forms an acidic solution and should be buffered before use
- Injectable agents are useful in larger fish, but careful dosing is required and recovery times are often longer
- Local analgesia may be performed, with care not to overdose small fish
- Euthanasia may be carried out by overdose of waterborne agents, injectable agents or percussive means. Clove oil may be used by owners for euthanasia of fish

INDUCTION AND MAINTENANCE OF ANAESTHESIA

Sedation

Most procedures in fish requiring restraint are usually carried out using anaesthetic agents at the appropriate plane of anaesthesia. Premedicant sedatives are not usually employed because of the adverse effects of the extra handling required in giving parenteral drugs.

Mild hypothermia will tranquillise or immobilise fish (Bell, 1964). This can be achieved by refrigeration, adding ice to the water or the use of solidified carbon dioxide (this should not come into contact with the water) (Ross, 2001). Care should be taken not to lower the temperature too far or too quickly, and it should be remembered that some tropical species may not tolerate even limited temperature

decreases. Hypothermia almost certainly gives little analgesia and so should not be used for potentially painful procedures (Ross and Ross, 1999).

Sedation may be useful in long distance fish transportation where the resulting reduction in metabolic rate and waste excretion can improve water quality and dissolved oxygen levels (Ross, 2001). Eugenol sedation has been shown to suppress circulating cortisol levels during crowding and low oxygen conditions (Small, 2004), and so sedation may reduce transport stress. Moderate cooling, reduced doses of waterborne anaesthetic agents (such as MS-222 and benzocaine) or a combination of both have been used.

Some species of elasmobranchs will go into a state of tonic immobility when turned on their backs (George, 2004; Harms, 2003). This technique can be employed for minor procedures, such as taking blood samples; however, a strong stimulus may arouse the animal.

Induction

As discussed, waterborne anaesthesia is the most widely used anaesthetic modality for fish (Harms, 1999) and usually the most appropriate for the practice setting. The fish to be anaesthetised is placed in an induction tank containing the anaesthetic solution. To minimise stress to the fish, this solution should ideally be water from the fish's usual environment to which the anaesthetic agent has been added. The owner should be advised to bring enough water to make up the induction solution, along with a similar volume into which the fish can be placed to recover.

This induction tank should be large enough to accommodate the fish comfortably, including sufficient water depth, but should not be too large to allow the fish to damage itself in any excitement phase of anaesthesia. A plastic storage container (made of non-toxic plastic), without any sharp edges, is a good choice for most fish (Fig. 18.9). The plastic transport bag may be used, but care should be taken that the fish does not become trapped in a corner as this may impair ventilatory effort. Because of the possible excitement phase during induction and the irritant properties of some agents, it is advisable to have a lid on induction and recovery tanks as some fish may jump out of the water.

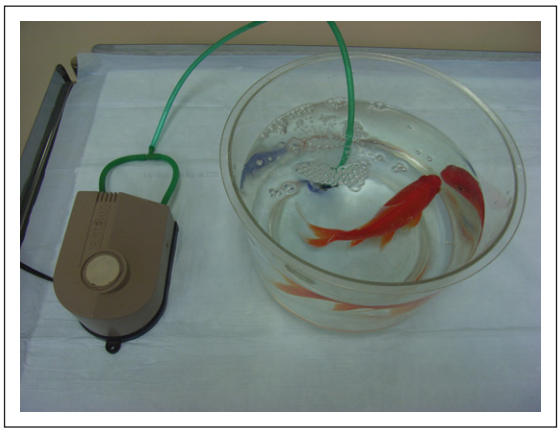


Figure 18.9 • A goldfish (*Carassius auratus*) in a suitable induction tank. The tank should be large enough to accommodate the fish comfortably, but small enough to prevent trauma in any excitement phase. Note the air pump, connected to an airstone, aerating the water.

As many anaesthetic agents tend to cause respiratory depression, the induction chamber should be aerated. This is best achieved using an airstone connected to an air pump designed for aquarium use, available from pet stores. Administration of pure oxygen may be used; however, prolonged exposure to excessively high oxygen concentrations can damage the gills (Harms, 1999). Oxygen concentration should be maintained between 6 and 10 ppm (Harms, 1999), although in most situations it is not necessary to measure this. Oxygen-supersaturated water may in itself cause narcosis (George, 2004).

Ideally induction should be rapid to reduce stress and reduce the duration of any excitement phase. This is best achieved by using a higher dose of anaesthetic agent that can then be reduced for maintenance. Some dosages are given (see Table 18.6), but it should be stressed that these are guidelines only. When using a novel agent or dealing with an unfamiliar species it may be more prudent to start at lower concentrations and increase the dose gradually to effect. If working with a number of fish it may be best to trial the anaesthetic regime on a small number of fish first and monitor for delayed adverse effects over the following 12–24 h before proceeding with anaesthesia of the remainder of the fish (Brown, 1988).

Maintenance

For short procedures (under 5 min) the fish may be simply removed from the solution and either returned to the anaesthetic solution to extend the anaesthesia or placed in a recovery tank free of anaesthetic solution to recover. Fish should not spend extended periods in the induction tank.

For longer procedures, which necessitate the fish being out of water, water needs to be directed over the gills to allow respiratory gas exchange and maintenance of anaesthesia. This can be achieved by using either a non-recirculating or a recirculating system.

Non-recirculating systems are the simplest to construct, require less equipment and work well for small fish. Following removal from the induction chamber, the fish is placed on a surface that will not damage the cuticle (a waterproof drape or the reverse plastic coating of an incontinence pad are good choices). Water is directed over the gills and drains away. This can be achieved using a handheld syringe (see Fig. 18.6) or a turkey baster directed into the mouth so the water exits the opercular openings. An intravenous fluid bag connected to a standard giving set filled with anaesthetic solution can also be used to direct water over the gills (Fig. 18.10). The control valve can be used to regulate the flow. Unrestricted flow will produce about 250 ml/min, which is enough for a 250 g fish (Ross, 2001). Like the induction chamber, the fluid bag should be aerated using an airstone.

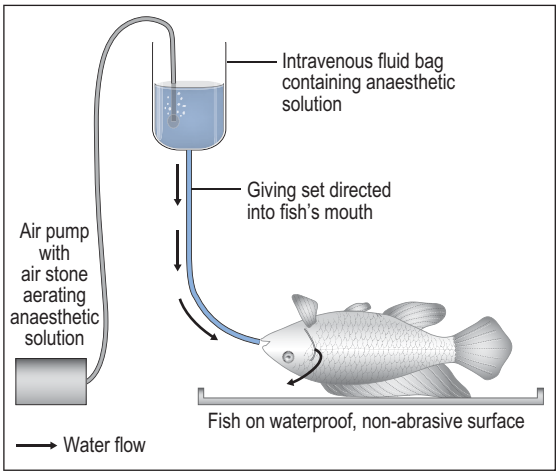


Figure 18.10 • A non-recirculating anaesthetic system. The fish is placed on a non-abrasive surface. An intravenous fluid bag is filled with anaesthetic solution and the tubing from the giving set is directed into the fish's mouth. The flow of water into the mouth (and then over the gills) can be controlled by adjusting the valve on the giving set.

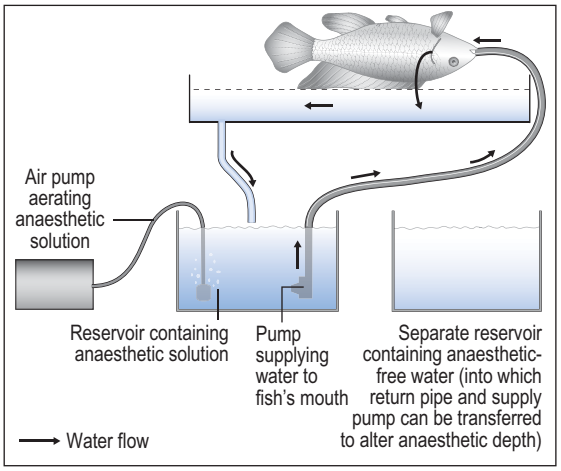


Figure 18.11 • A recirculating anaesthetic system. An electric pump is positioned in a reservoir tank containing anaesthetic solution. Tubing from the pump is directed into the fish's mouth. The anaesthetic solution runs off from the fish and drains back into the reservoir tank. A separate reservoir with anaesthetic-free water can be positioned in the system – anaesthetic depth can be varied by switching the pump and return pipe to this reservoir.

Changing anaesthetic depth can be achieved by directing water containing different anaesthetic concentrations over the gills, dependent on assessment of the anaesthetic depth of the patient. It may be sensible to prepare in advance several 60 ml syringes or fluid bags containing higher anaesthetic concentration water and anaesthetic-free water so that changes may be effected quickly (Harms, 2003).

Recirculating systems deliver anaesthetic solution across the gills that is then collected and returned to the fish repeatedly. This system is more economical for larger fish.

These systems can be most simply achieved by modifying the non-recirculating systems above. The fish is placed over a reservoir that collects the water that runs out of the gills. Instead of using fresh anaesthetic solutions directed over the gills, the operator returns water collected by hand to the fish by use of the syringe or by refilling the fluid bag and giving set arrangement. A less labour-intensive solution is to use an electric pump to return water to the gills from the reservoir (Fig. 18.11). This method is preferable for longer procedures. Electrical safety for the operator should be considered and the use of low voltage pumps or a residual current device (RCD) employed.

Tubing from the electric pump is placed in the fish's mouth (Fig. 18.12). The flow rate may be controlled by opening and closing a valve that directs a proportion of the water directly back to the reservoir. Flow rates of 1–3 L/min have been successfully used (Harms, 1999). If the rate is too slow then there will be poor gas exchange. A flow that is too fast may damage the gills or cause gastric inflation. Use of soft tubing, ideally silicon rubber (Ross, 2001), will reduce the risk of damage to the buccal cavity. Care should be taken not to insert the tubing too caudally as this may result in gastric inflation rather than

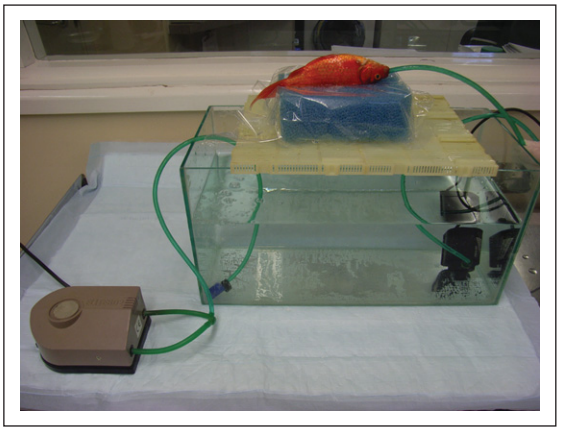


Figure 18.12 • A recirculating anaesthetic system in operation. Note the electric pump within the reservoir with tubing to the fish's mouth. The airpump on the left is aerating the water.

effective water flow over the gills (S. Billington, unpublished work, 2005). Again it is important to aerate the water reservoir to increase oxygen concentration and also to reduce dissolved carbon dioxide.

Changes in anaesthetic depth may be achieved by altering the water anaesthetic concentration. A second reservoir containing anaesthetic-free water can be positioned in the system so that if the anaesthetic depth needs to be changed the pump and return pipe from the work surface drain can be switched between the reservoirs. This gives the option of interchangeably supplying the fish with anaesthetic solution or anaesthetic-free water.

Fish may be maintained on such a recirculating system for several hours. For longer procedures, or when solutions are reused for large numbers of fish, attention should be paid to maintaining water quality. The main factors requiring control are temperature, dissolved oxygen concentration, ammonia levels and build-up of faecal solids (Ross and Ross, 1999). It should also be remembered that the solution will gradually become anaesthetic-depleted.

To maintain water temperature, a submersible aquarium combined heater and thermostat can be placed in the reservoir. When water quality starts to deteriorate changing of the solution will be required; timing for this can be determined by observation of longer induction times in subsequent fish or by direct measurement of water parameters (Harms, 1999). Increasing protein concentration from fish cuticle will cause increased foaming of aerated water and this can be used as a visual indication of deteriorating water quality.

Whenever the fish is kept out of the water the skin should be kept moist to prevent damage. Surgical drapes over the fish may offer some protection from desiccation and overheating due to surgical lights.

Recovery

On completion of the required procedure, the fish is placed in anaesthetic-free water, which should be well aerated and match the tank or pond conditions. During recovery the light levels should be dimmed to reduce stress. It may also be preferable to release the fish in the water before full equilibrium has returned as further handling may precipitate a more stressful recovery. Respiration, motion and equilibrium should be monitored during this time (Harms, 1999).

BOX 18.5 Induction and maintenance of anaesthesia

- Sedation and general anaesthesia are relative stages on a continuum, dependent on dose of anaesthetic agent and length of exposure (Ross, 2001)
- Injectable premedicants are not usually used before anaesthetic induction
- Induction tanks should be aerated as many waterborne agents cause hypoxia
- For extended periods out of the water, water should be directed over the gills (from the mouth) to maintain respiratory gas exchange and, with waterborne agents, maintain anaesthetic depth. Syringes, intravenous fluid bags (with giving sets attached) or pumps may be used for this
- With waterborne agents, changes in anaesthetic depth are carried out by changing the anaesthetic concentration of the solution directed over the gills. Recovery is achieved by placing the fish in anaesthetic-free water or directing this water over the gills

ANAESTHESIA MONITORING

Planes of anaesthesia

The stages of fish sedation and anaesthesia are shown (Table 18.3; Fig. 18.13). These stages are dependent on the dose rate of the anaesthetic agent and length of exposure (Ross, 2001). At high concentrations of induction agent, the fish may pass rapidly through these individual stages and in practice the stages may be indistinct.

For short procedures, anaesthetic depth may be determined by the response to tactile and surgical stimuli and the respiratory rate, which is seen as opercular movement (see Table 18.3; Fig. 18.13) (Harms, 2003). The depth of anaesthesia required will depend on the nature of the procedure performed. For full surgical anaesthesia, the respiratory rate may be very slow (Brown, 1993) and so other means of monitoring should also be employed. For more involved or prolonged procedures, other techniques should also be utilised to assist monitoring of the anaesthetic.

Cardiovascular system

Measurement of heart rate is useful in surgical cases and also during longer procedures. Pulse is not readily palpable in fish (Harms, 2003), but in some fish the heart can be observed beating though the skin on the ventral midline caudal to the base of the operculae (Lewbart, 2001a). Use of a Doppler ultrasound probe is usually practicable and in most fish can be positioned over the heart for audible cardiac monitoring (see Fig. 18.7).

Heart rate will vary between species and with stress and temperature, but normal rates (in unanaesthetised fish) are often between 30 and 70 beats per minute (Lewbart, 2001a). Cardiac ultrasonography and an ECG may also be performed (Harms, 2003). ECG electrodes may be clamped lightly on to the pectoral and anal fins (Ross, 2001) or subcutaneous leads used.

Blood gases can be measured in fish of a sufficient size (Harms, 2003). Pulse oximetry has not shown consistent results (Ross, 2001) and is not recommended as a means of anaesthetic monitoring.

Respiratory system

Slow and regular opercular movements, with minimal response to manual stimuli, indicate sufficient depth of anaesthesia for most procedures. When placed in air, opercular movements may be reduced (Brown, 1993).

Table 18.3: Stages of anaesthesia in fish

STAGE	PLANE	CATEGORY	BEHAVIOURAL RESPONSE OF FISH					COMMENTS
			Voluntary swimming	*Reaction to visual and tactile stimuli*	*Respond to postural changes*	*Muscle tone*	*Respiration rate*	
0		Normal	Yes	Yes	Yes	Normal	Normal	
I	1	Light sedation	Yes	Slight decrease	Yes	Normal	Normal	
I	2	Deep sedation	No	Decreased – response to strong tactile stimuli present	Yes	Slightly decreased	Slightly decreased	
II	1	Light narcosis	No	Decreased – still response to strong tactile stimuli present	Decreased	Decreased	Increased	
II	2	Deep narcosis	No	Very decreased – weak response to strong tactile stimuli present	No	Decreased	Normal	Suitable for external sampling, fin biopsies, gill biopsies
III	1	Light surgical anaesthesia	No	Very decreased – weak response to strong pressure present	No	Further decreased	Decreased	Suitable for minor surgical procedures
III	2	Surgical anaesthesia	No	No	No	None	Very decreased	
IV		Medullary collapse	No	No	No	None	Absent	Followed in several minutes by cardiac arrest

(Adapted from Brown, 1993)

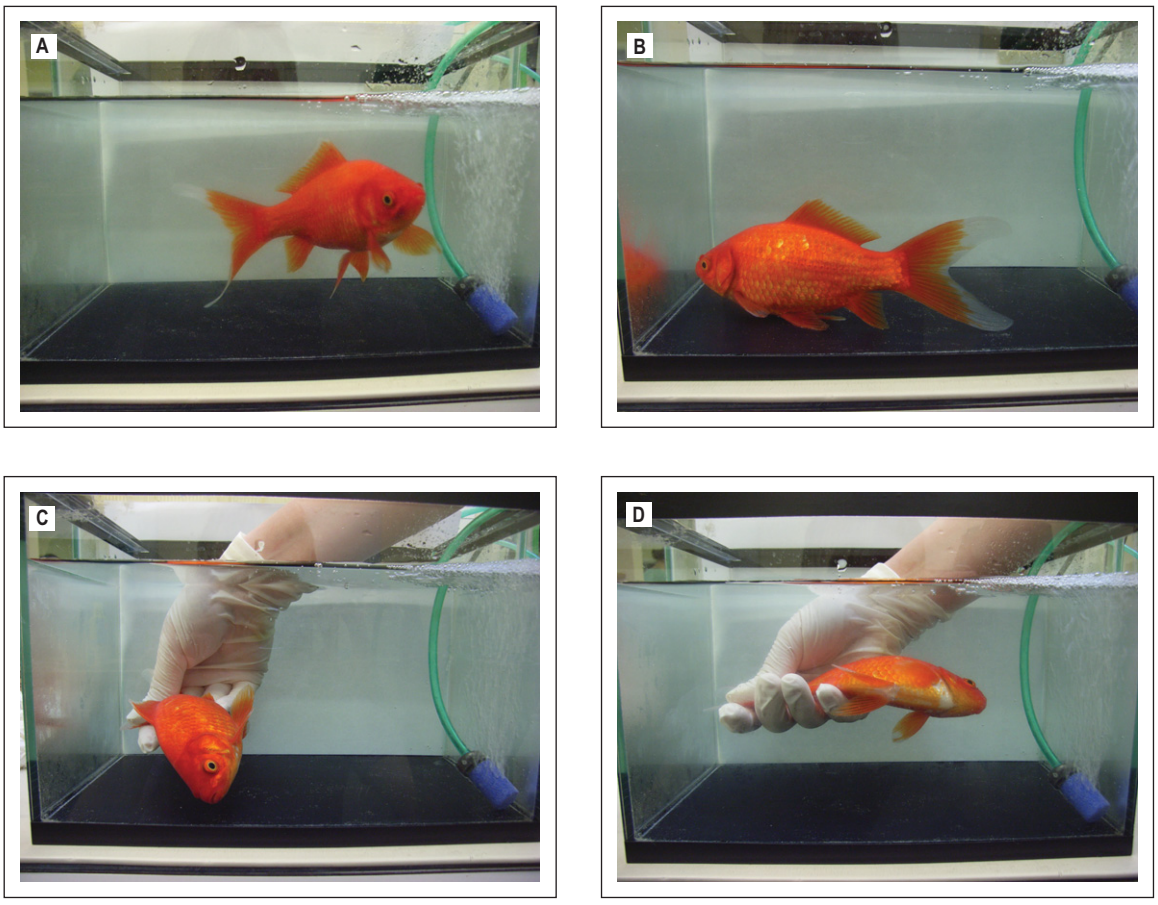


Figure 18.13 • Stages of fish anaesthesia induced by gradual increasing water concentrations of MS-222. (A) Stage 0 – normal. Voluntary swimming, normal equilibrium and responsive. (B) Stage I.2 – deep sedation. Not swimming, normal equilibrium and decreased response to stimuli. (C) Stage II.2 – deep narcosis. Very decreased response to tactile stimulus and loss of response to postural changes. (D) Stage III.2 – surgical anaesthesia. No response to strong tactile/surgical stimulus.

Central nervous system

Jaw tone, which may be present in the absence of opercular movement, may be used as an indicator of anaesthetic depth (Harms, 2003).

BOX 18.6 Monitoring

- For simple procedures anaesthetic depth may be monitored by response to tactile/surgical stimuli and respiratory (opercular) rate
- More involved procedures should involve monitoring heart rate, visually or by Doppler probe
- Electrocardiogram (ECG), cardiac ultrasonography, jaw tone and blood gases may also be used
- At surgical anaesthetic planes, respiratory rate may be very slow

PERI-ANAESTHETIC SUPPORTIVE CARE

Analgesia

As discussed earlier, there is debate as to the perception of pain in fish. However, various studies have demonstrated the possibility that fish experience pain (Chandroo et al., 2004; Dunlop et al., 2006; Sneddon, 2003a) and so it would appear justifiable to use analgesics in potentially painful situations. Opioids, non-steroidal anti-inflammatory drugs (NSAIDs) and local anaesthetics may be used in fish (see Table 18.4) (Harms, 2003; Harms et al., 2005). Intraoperative use of butorphanol in koi (*Cyprinus carpio*) was shown to reduce post-surgical negative behaviours (Harms et al., 2005). Studies suggest that intraoperative use of ketoprofen had no behavioural sparing effect, but the anti-inflammatory action may result in less muscle damage following surgery (Harms et al., 2005).

FORMULARY

Table 18.4: Suggested doses of analgesics used in fish

DRUG	DOSE (mg/kg IM)	COMMENTS
Butorphanol	0.1–0.4[2]	Analgesic[1]
Ketoprofen	2[1]	Anti-inflammatory[1]

Key: IM = intramuscular
[1] (Harms et al., 2005); [2] (Lewbart, 2001c)

Table 18.5: Emergency drugs for use in fish

DRUG	DOSE	COMMENTS
Doxapram	5 mg/kg prn IV, IP[2]	Sharks Treatment of respiratory depression[2]
Atropine	0.1 mg/kg prn IM, IP, IV[1]	
Adrenaline (epinephrine) 1:1000	0.2–0.5 ml IP, IM, IV, IC[2]	Cardiac arrest[2]

Key: IM = intramuscular, IP = intraperitoneal (intracoelomic), IV = intravenous, IC = intracardiac, prn = 'as the situation arises'
[1] (George, 2004)[2] (Stoskopf, 1993b)

Drug metabolism will be temperature-dependent and so this should be factored into dosing interval calculation. In a study of the pharmacokinetics of morphine there was a wide species variation even at the same temperature, although the disposition of morphine in fish was approximately one order of magnitude slower than it is in mammals (Newby et al., 2005).

EMERGENCY PROCEDURES

Resuscitation

If opercular movements stop, particularly if the fish is in water, or there are other signs that the anaesthetic depth is too great, forced recovery/resuscitation efforts should be made (Harms, 2003). Reversal agents to any injectable anaesthetic previously administered should be given, for example atipamezole to reverse alpha-2 agonists. In preference the fish should be placed in the recovery tank and anaesthetic-free water directed across the gills. This may

Table 18.6: Anaesthetic drug doses in fish

AGENT	DOSE (concentration in water or injectable dose rate)	COMMENTS
Waterborne agents		
MS-222, tricaine methanesulfonate	100–200 mg/ml[5] 50–100 mg/ml[5] 15–50 mg/ml[5] 1g/L spray[1]	Induction Maintenance Sedation Large fish Spray on to gills with aerosol sprayer
Benzocaine	Approx. same doses as MS-222[10] 25–200mg/ml[9]	
Eugenol (clove oil)	25–50 mg/ml[11] More than 10 drops/L[9]	Anaesthesia Euthanasia
Eugenol/ Polysorbate 80 (Aqui-S®, Aqui-S, New Zealand Ltd)	17–25 mg/ml[12] 6mg/ml[12]	Anaesthesia Sedation
Quinaldine sulphate	50–100 mg/ml[5] 15–60mg/ml[5]	Induction Maintenance
Metomidate	0.06–0.2 mg/ml[5] 0.5–1 mg/ml[5] 2.5–5 mg/ml[5] 5–10 mg/ml (up to 30 mg/ml for some species)[5]	Transport Light sedation Heavy sedation Anaesthesia Some authors recommend much lower doses Gouramis very sensitive[5]
Isofluorane/ halothane	0.5–2 ml/L[5]	Added directly to water, or vaporise and dissolve to effect Other agents preferable
Carbon dioxide gas	Bubble gas through water[8]	Euthanasia
Sodium bicarbonate	30 g/L[8]	Euthanasia
Alka Seltzer®, Bayer	2 tablets/0.5–1.0 L3	Euthanasia

Continued

Table 18.6 (Continued)

AGENT	DOSE (concentration in water or injectable dose rate)	COMMENTS
Ethanol	> 3% in water[4]	Euthanasia; other agents preferable[7]
Injectable agents		
Ketamine	66–68 mg/kg IM[9]	Teleosts (bony fish)
	12–20 mg/kg IM[9]	Elasmobranchs (sharks and rays)
Ketamine/medetomidine	1–2 mg/kg ketamine with 0.05–0.10 mg/kg medetomidine IM[5]	Teleosts; reverse with 0. 2 mg/kg atipamezole IM.[5]
Ketamine/xylazine	12–20 mg/kg ketamine with 6 mg/kg xylazine IM[5]	Sharks
Alfaxolone-alphadalone (Saffan)	12 mg/kg IV[9]	
Propofol	3.5–7.5 mg/kg IV[2]	
Pentobarbital	60 mg/kg IP[8]	Euthanasia
Lidocaine (lignocaine)	Do not exceed 1–2 mg/kg total dose[5]	Local anaesthesia[5]

Key: IM = intramuscular, IP = intraperitoneal (intracoelomic) IV = intravenous

[1] (Brown, 1993); [2] (Fleming et al., 2003); [3] (Gratsek et al., 1992); [4] (Harms, 1999); [5] (Harms, 2003); [6] (Lewbart, 2001c); [7] (Lewbart, 2005); [8] (Noga, 2000); [9] (Ross, 2001); [10] (Ross and Ross, 1999); [11] (Sladky et al., 2001); [12] (Treves-Brown, 2000)

be achieved by facing the fish into the flow of the filter/aerator or by holding the fish and moving it forwards slowly though the water, ideally whilst holding the mouth open. Fish should not be dragged backwards through the water as the reverse flow of water may damage the gills, as well as greatly reducing the efficacy of gas and anaesthetic exchange (Brown, 1988). A syringe or rubber enema pump may also be used to direct water through the mouth (Ross, 2001). Resuscitation attempts should not be abandoned prematurely as respiratory arrest can precede cardiac arrest by an extended period of time, possibly several minutes. Once spontaneous ventilation returns, recovery should proceed as above.

Emergency drugs

In the case of respiratory arrest being unresponsive to the resuscitation procedures outlined above, or in the case of cardiac arrest, the use of respiratory and cardiac stimulants may be appropriate (Table 18.5). Intravenous administration of doxapram to sharks immobilised with ketamine and xylazine produces dramatic arousal (Stoskopf, 1993d).

Acknowledgements

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19 Invertebrate anaesthesia

Michelle O'Brien

INTRODUCTION

The invertebrates are an extremely diverse group of animals (see Table 19.1). Many are highly specialised, with differing body shapes, methods of feeding and number of legs being just some of the differences that separate the different groups. All invertebrates lack a backbone. This in turn means that most are noticeably smaller and many are looked upon as more 'fragile' than many other species of animals.

The simplest means of discussing anaesthesia in these species is to separate the procedures used into those for aquatic and those used for terrestrial invertebrates. In certain circumstances the same agents can be used both in water and in air, for example, gaseous anaesthetic agents can be bubbled though water in order to anaesthetise aquatic invertebrates. In most cases injectable agents are not used in invertebrates due to their small size, the chance of leading to iatrogenic damage and the difficulty in correct dosing. In this chapter we will be concentrating on the invertebrate species most commonly encountered in general practice, but will also try to touch on some aspects of anaesthesia for some more unusual specimens (Figs 19.1–19.3).



Figure 19.2 • Blue crayfish (*Procambarus alleni*).



Figure 19.1 • Atlantic anemones (*Condylactis* sp.).



Figure 19.3 • Red starfish (*Fromia milleporella*).

Some invertebrates, for example arthropods and molluscs, have been shown to have both the body wall and internal tissues well endowed with nerve endings or similar sensory structures (Cooper, 1998) and, although invertebrate pain is a topic for debate, it is wise to err on the side of caution. In countries where animal welfare issues are of high priority it is becoming clearer that invertebrates, like any other animal, should be given the benefit of the doubt (Cooper, 2006). Analgesia has not been studied in most invertebrates, but the adverse response to noxious stimuli in many species suggests that anaesthesia should be employed in any situation where painful procedures are to be undertaken. Anaesthesia is useful for haemolymph sampling, oral swabs and flushing of the mouthparts for parasite evaluation in spiders (Pizzi, 2006), together with any examination of venomous specimens or delicate procedures requiring complete immobilisation of the individual.

In a number of invertebrate species surgery such as sectioning and grafting is carried out without any form of anaesthetic, for example in sponges and tubellarians. In coelenterates (a term encompassing both phylum Cnidaria and comb jellyfish), due to questions regarding their ability to feel pain, anaesthesia is employed more for control of motion to facilitate examination, imaging or manipulation than for analgesia. Many procedures are carried out without anaesthesia, especially in sessile species (Stoskopf, 2006).

ANATOMY AND PHYSIOLOGY

The most important factor in dealing with any invertebrate species is knowledge of exactly which species you are treating and its environmental requirements and behavioural patterns (Table 19.1). An example of this is theraphosid spiders (tarantulas) that will turn over into dorsal recumbency in specially designed webs during moulting. Any disturbance or righting of the animal may lead to disruption of the moult and can be fatal.

Many invertebrates have multiple, relatively delicate, limbs or wings and, therefore, must be treated with caution to prevent accidental injury. The opisthosoma (abdomen) is very delicate and can easily be damaged by falls, often fatally; therefore, experience and care in handling are essential.

Table 19.1: Some of the significant taxonomic classifications of the invertebrates dealt with in this chapter

PHYLA	CLASSES, SUBCLASSES AND ORDERS	COMMON NAME
Porifera		Sponges
Cnidaria	Class Anthozoa (anemones, corals, etc.) Class Cubozoa (sea wasps or box jellyfish) Class Scyphozoa (cup animals and jellyfishes)	Sea anemones, jellyfishes, corals, hydroids
Echinodermata	Class Asteroidea (sea stars) Class Echinoidea (echinoids, sea urchins and sand dollars) Class Holothuroidea (sea cucumbers) Class Ophiuroidea (brittle stars and basket stars)	Sea urchins, starfish, brittle stars, sea cucumbers
Mollusca	Class Bivalvia (clams, mussels, oysters, scallops) Class Cephalopoda (squids, octopus, nautilus, cuttlefish) Class Gastropoda (snails, sea slugs)	Molluscs (slugs, snails, squid)
Annelida	Subclass Hirudinea (leeches) Subclass Oligochaeta (angleworms, earthworms and their relatives, night crawlers, and oligochaetes) Class Polychaeta (paddle-footed annelids and polychaetes)	Segmented worms
Arthropoda	Subphylum Chelicerata, class Arachnida (arachnids) Subphylum Crustacea (crustaceans), class Malacostraca, order Decapoda (crabs, lobsters, etc.) Subphylum Hexapoda, order Diplura (diplurans), class Insecta (insects) Subphylum Myriapoda, class Chilopoda (centipedes), class Diplopoda (millipedes)	Arthropods (insects, crabs, spiders, etc.)

Cardiovascular system

In spiders the heart lies in the middorsal opisthosoma (Fig. 19.4). Demarcation of organs is often inexact and most are embedded between the multiple midgut diverticula that occupy most of the opisthosoma (Pizzi, 2006).

The circulatory system of spiders involves a well-developed arterial and open venous circulation, with haemocyanin the copper-based oxygen carrying pigment (Pizzi, 2006). Vessels from the heart direct haemolymph to specific organs and throughout the spider's body. Venous return follows specific pathways and occurs due to a pressure gradient. The pericardial sinus around the heart, together with ligaments attached to the cuticle, helps generate suction and pressure. Haemolymph is clear with a blue tinge due to the haemocyanin. Arachnids need to maintain blood volume and pressure for leg extension; therefore, dehydrated individuals often hold their legs flexed beneath them and are unable to move. Haemocyanin has a much higher oxygen affinity than haemoglobin and functions more as an oxygen storage mechanism than a transporter. Spiders have a very low metabolic rate with an oxygen consumption of one-hundredth that of warm-blooded vertebrates and one-fifth that of poikilothermic vertebrates (Paul, 1990).

The resting heart rate can range from 30–40 beats per minute in large theraphosids, to over 100 beats per minute in smaller spiders. The rate can double after short bursts of activity (Foelix, 1996). One study (Coelho and Amaya, 2000) found the resting heart rate of the Texas tarantula (*Aphonopelma hentzi*) to be 5.6 ± 1.5 beats per minute.

Owing to a hard exoskeleton and an open or mixed circulatory system (Noga et al., 2006), surgery is usually not feasible in crustaceans. It is, however, possible to use injectable anaesthetics, though only at the level of the joints where there is a thinning of the exoskeleton termed the arthrodial membrane (Fig. 19.5) (Oswald, 1977).

Owing to an open or semi-closed circulatory system in many invertebrates, any damage to the exoskeleton can lead to excessive haemorrhage and death by exsanguination.

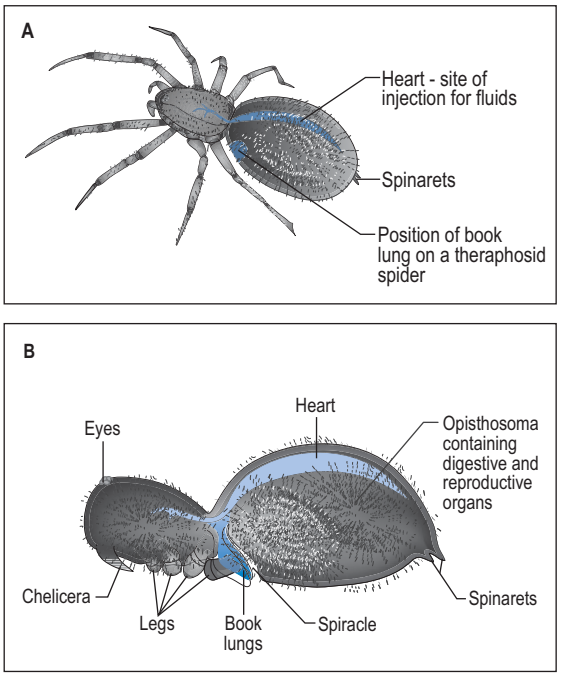


Figure 19.4 • Position of the heart and book lungs in a theraphosid spider. (A) Spider. (B) Detail.

Respiratory system

The folia (leaves) of the book lungs are present in the cranio-ventral opisthosoma. Spiders' book lungs on the ventral abdomen enable anaesthesia using gaseous agents by having only the abdomen within the gas chamber. This leaves the cephalothorax readily accessible for assessment and procedures (G. A. Lewbart, unpublished work, 2006). Many spiders also have trachea that open above the spinnerets. Gases are exchanged with the blood by diffusion. Book lungs hang in an open space connected to a tube. The other side of the tube is connected to the open air. Most spiders possess both book lungs and trachea, though some only possess one mechanism. The trachea supplies oxygen faster than the book lungs. More 'modern' spiders (for example, the family Symphyltognathidae) that only possess tracheal systems, therefore, have quicker and longer reactions than more primitive species that only possess book lungs (Nieuwenhuys, 2006).

Crustaceans use gills for respiration and these are also the main site for absorption of aqueous anaesthetic agents (Noga et al., 2006).

Integumentary system

Invertebrates are a highly diverse group of animals that vary greatly in the risks they can pose to humans (Fig. 19.6). These risks include toxic secretions (for example, venom); urticarial hairs on the abdomen of the theraphosid spiders (Fig. 19.7); painful bites, which often also act to transmit bacteria; stings and defensive spines (Cooper, 2001).

Some species can also shoot venom at a distance, for example Assassin bugs (family Reduviidae), and goggles

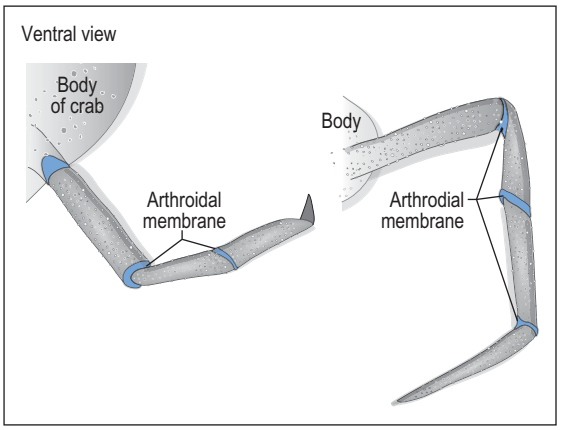


Figure 19.5 • Arthrodial membrane position in the leg of a decapod.



Figure 19.6 • It is advisable to wear thick gloves when handling *Heteropteryx* sp. stick insects due to large spines on their hind legs.



Figure 19.7 • Abdomen of a Baboon spider (*Pterinochhilus murinus*) showing urticarial hairs. (Photo courtesy L Longley)

should be worn when dealing with these species. The production of risk assessments can be beneficial in a veterinary practice dealing with these species.

Nervous system

Crustaceans use 5-hydoxytryptamine glutamate and possibly peptide-like substances as well as the more common forms of neurotransmitter. They can, therefore, respond differently to certain anaesthetic substances compared to other species, possibly because their synaptic receptor sites are not affected (Ross and Ross, 1999). The reflex grasping action of many decapods can lead to a type of immobilisation wherein simple procedures, for example fitting of near-heart electrodes, can be accomplished during this distraction.

Many agents other than magnesium chloride, which is thought to act on the central nervous system, are ineffective in cephalopods. This may be due to the nature of their nerve transmitters and receptors (Messenger et al., 1985).

Owing to a difference in neurotransmitters, crustaceans can react differently to anaesthetics than other invertebrates.

PRE-ANAESTHETIC ASSESSMENT/STABILISATION

History

Prior to anaesthesia it is useful to have a full assessment of the environment previously inhabited. Invertebrates are ectothermic and unable to control their body temperature other than by behavioural modification. Temperature and humidity are, therefore, vitally important, especially regarding correction of dehydration prior to anaesthesia.

Clinical examination and further assessment

Depending on the species, a visual inspection or hands-on examination may be performed to assess the patient's condition.

It is important to determine secondary problems present other than those that anaesthesia is required to correct. These can include parasitic, fungal and bacterial infection, or the close proximity of some individuals to moulting (this process could be disrupted by anaesthesia and if interrupted can lead to fatalities). In theraphosid spiders the colour of the opisthosoma may darken prior to moulting. This is especially visible in areas of alopecia. The spider will also become anorexic prior to moulting (Pizzi, 2006).

In aquatic invertebrates water quality should be maintained by oxygenation and filtration, with temperature maintained to prevent overheating. Maintenance of a damp skin or exoskeleton for aquatic invertebrates is a vital process (Ross and Ross, 1999) and can be important in some terrestrial invertebrates with delicate cuticles, for example myriapods. For this reason, the carrier gas should be humidified (Chitty, 2006).

During anaesthesia temperature and humidity must be maintained for terrestrial invertebrates, together with water quality for the aquatic species.

All pre-anaesthetic assessments should include the question of whether a general anaesthetic is necessary or appropriate for the procedure to be carried out – for example, in spiders autotomy of limbs is a voluntary process and, therefore, must be carried out without anaesthetic (Pizzi, 2006).

Autotomy of limbs in spiders is a voluntary process and, therefore, anaesthesia must not be administered.



Figure 19.8 • A Huntsman spider (*Heteropoda venatoria*) provided with a dish containing soaked cotton wool to drink from, as a means to combat dehydration.

Fluid administration

Increasing spraying or providing a dish of water from which the individual can drink (Fig. 19.8) can improve the outcome of the anaesthetic and procedure carried out.

It is possible to provide fluids via injection (using a 26 or 27 gauge needle) in certain species, for example theraphosid spiders. 0.9% sodium chloride, lactated Ringer's solution and Hartmann's solution have all been administered to theraphosid spiders without incident (Pizzi, 2006). It is also possible to prepare a spider Ringer's lactate to approximate the composition of tarantula haemolymph (Schartau and Leidescher, 1983).

BOX 19.1 Recipe for spider Ringer's lactate (Schartau and Leidescher, 1983)

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Please refer to the printed publication.

In a conscious spider fluids can be administered into the ventral aspect of a limb joint, through the arthrodial membrane, with the spider restrained in dorsal recumbency using a plastic ruler. Even if damage to the limb is sustained it can be removed using autotomy and the stump sealed using cyanoacrylate glue (Pizzi, 2006). However, administration is slow and only a limited amount of fluid can be administered.

Johnson-Delaney has been quoted (Chitty, 2001) as recommending a different method of fluid administration. In an anaesthetised or severely dehydrated spider fluid can be administered into the heart or pericardial sac present in the dorsal opisthosoma. Administration is rapid and a relatively large amount of fluid can be given at one time. Owing to the open haemolymph system, extracardiac administration because of incorrect placement is not detrimental. However, if the spider or fluid administrator were to move during the procedure, fatal cardiac laceration could result (Pizzi, 2006). A suggested fluid administration amount is up to 20 ml fluid/kg body weight. This amount should be reduced depending on variation in weight due to the amount of ingesta present (Johnson-Delaney 2000).

In scorpions that are too severely dehydrated to drink when water is offered in a shallow dish, a similar administration can be performed. An appropriate volume of sterile half-strength physiological dextrose saline can be injected dorsolaterally between the platelike tergites of the mesosoma (preabdomen). A 30 gauge, or smaller, needle attached to an insulin syringe should be used and the needle advanced only deep enough to permit injection but not so deep as to risk injury to internal organs (Frye, 2006).

Myriapods also require fluid therapy in certain cases; however, systemic administration has yet to be evaluated. Millipedes may be placed in a shallow bowl of lukewarm water and centipedes may be placed in a very humid environment surrounded by moistened tissue paper (Chitty, 2006) in order to treat clinical dehydration.

Many arthropods will often take fluid offered directly from the tip of a syringe. In other species spraying and providing soaked cotton wool may be sufficient. In molluscs and aquatic arthropods rehydration can be accomplished by immersion or soaking in an appropriate solution, depending on the species (Cooper, 1998).

EQUIPMENT REQUIRED

The majority of invertebrate anaesthetics do not require any specialist equipment that is not present in a general practice that deals with small mammals. Most of the anaesthetics for terrestrial invertebrates involve placing the animal in an anaesthetic chamber and administering gas anaesthetic. The most vital piece of equipment is, therefore, the anaesthetic chamber that can be created in a number of different ways – varying from using a small animal facemask to contain the animal, to using a purpose-built small mammal anaesthetic chamber.

External sources of heat, for example heat mats or water-filled gloves – protected with thin towels from any damage by external projections from the specimen (such as spines in jungle nymph stick insects [*Heteropteryx sp.*] [see Fig. 19.6]) – are important for terrestrial invertebrates in order to reduce lowering of body temperature under general anaesthesia. It is also vital, given the size of the animal and in certain cases shape (for example, myriapods), that the ends of any accessible pipes or other apertures present in the anaesthetic chamber should be covered by fine mesh to prevent the patient gaining access to the anaesthetic circuitry (Chitty, 2006).

Smoke for bees requires specialist equipment and provides only a sedation effect, though it has been used for many centuries in order to facilitate the maintenance of hives. This is not a method regularly employed other than in specialist situations.



Figure 19.9 • Anaesthesia should not be performed within a mixed exhibit such as this marine tank, as many invertebrates may be affected.

The equipment required for aquatic invertebrates is usually a simple container holding a known volume of water either taken from the animal's previous container or made up as a fresh solution. It is important to ensure that temperature and salinity correspond to that of the animal's normal environment. In certain cases anaesthesia can be carried out in the tank in which the animal is normally housed, but this carries greater risks if other exhibits are housed in the same tank or if filtration will affect anaesthetic concentrations (Fig. 19.9) (Cooper, 2001).

TECHNIQUES

Handling

Handling and transferring of the animal from transport container to anaesthetic chamber can be facilitated by the use of nets, padded forceps in scorpions or direct handling using gloves in cooperative specimens with experienced handlers. In debilitated fragile specimens it can be easier to use a flat plastic scoop (Fig. 19.10) in order to reduce direct handling – this is especially true of specimens displaying dysecdysis. It is possible to use a gauze sling around the body between the cephalothorax and abdomen of theraphosid spiders while the forefinger applies gentle downward pressure on the cephalothorax to maintain restraint for certain procedures, by lifting them very slightly above a surface (Johnson-Delaney, 2000).

Those individuals capable of producing severe damage in a handler by using their defence mechanisms should be transferred only within suitable containers (Fig. 19.11) and anaesthetised with as little risk to the handler as possible. For example centipedes, *Scolopendra* sp. (Fig. 19.12), possess venom delivered in a bite known to cause excruciating pain; large theraphosid spiders can deliver a painful bite that may lead to bacterial infection or anaphylaxis. In venomous species, animals must not be handled by one person alone and antivenom should be available if possible – for example in black widow spiders (*Latrodectus mactans*). It is also important to note that this species, and a number of others, cannot be kept without licence in certain countries due to legislation, for example the Dangerous Wild Animals Act 1976 in the UK.



Figure 19.10 • Use of a plastic scoop to transfer a Chilean rose spider (*Grammostola rosea*) to the anaesthetic chamber.



Figure 19.11 • Use of a clear plastic container, available in practice, to transfer a Goliath bird-eating spider (*Theraphosa blondi*) to the anaesthetic chamber.



Figure 19.12 • Giant centipede (*Scolopendra* sp.), which can deliver an excruciatingly painful venomous bite. (Courtesy of L Longley.)

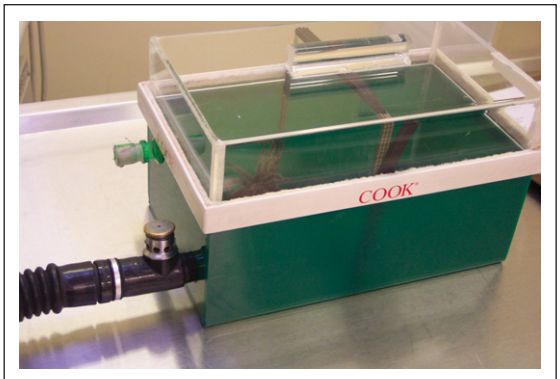


Figure 19.13 • Anaesthesia of a Chilean rose spider (*Grammostola rosea*) via gaseous anaesthetic in an anaesthetic chamber.

Induction and maintenance of anaesthesia

There are two main ways to anaesthetise a terrestrial invertebrate, such as a theraphosid spider. The first involves placing the spider into a small mammal anaesthetic chamber and administering gaseous anaesthetic, such as isoflurane, with the vaporiser set to maximum (4–5%) until anaesthesia is achieved (Fig. 19.13).

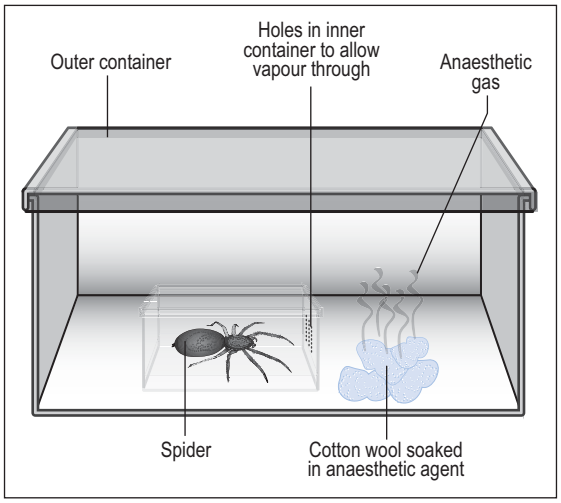


Figure 19.14 • The use of two containers as an anaesthetic chamber when a vaporiser is not available.

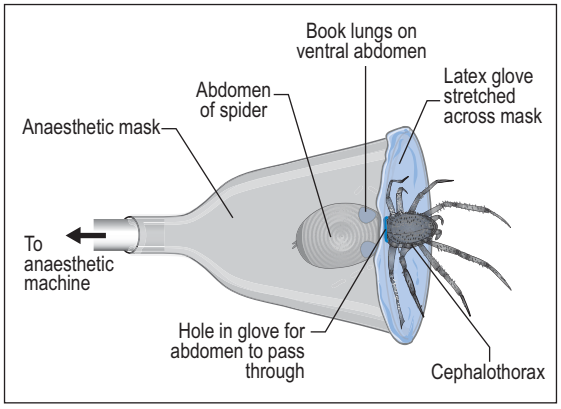


Figure 19.15 • A small animal facemask used as an anaesthetic chamber.

The second method involves placing the spider within a box with a number of holes in its surface within a second non-perforated container into which a ball of cotton wool soaked in gaseous anaesthetic agent is placed, thereby allowing no direct contact between the spider and the anaesthetic agent (Fig. 19.14). The spider is removed from the inner chamber once anaesthesia is sufficient – induction normally takes 10–15 min. It is possible to allow a degree of scavenging with the first system, but not with the second. The first should, therefore, be used if possible, due to health and safety concerns for staff regarding exposure to anaesthetic gases. The second method, however, can be carried out in circumstances in which no vaporiser is available (Pizzi, 2006).

Owing to the anatomy of spiders and the placement of the book lungs on their abdomens, through which anaesthetic is absorbed, it is possible to create an anaesthetic chamber using a small animal facemask (Fig. 19.15).

A latex glove is used to cover the open end of the mask and the abdomen of the spider is placed through a small hole in the latex, thereby lying inside the mask into which anaesthetic can be passed. This allows the cephalothorax to remain available for surgery and allows continuous anaesthesia, avoiding the necessity of returning the spider to the anaesthetic chamber using the previous methods if deepened or prolonged anaesthesia is required (G.A. Lewbart, unpublished work, 2006).

In decapods injectable anaesthesia can be carried out by injection into the haemocoel via the arthrodial membrane of a posterior leg (Fig. 19.5) or alternative route (Oswald, 1977). A number of anaesthetic agents have been given using this method, with varying results (see Table 19.5).

In an individual anaesthetised using aqueous solutions of agent, the animal is removed from the bath of solution after induction. Anaesthesia can be maintained by spraying the water containing the anaesthetic agent over the animal while it is lying on a damp surface. In order to lighten the anaesthesia oxygenated water without anaesthetic can be sprayed over the invertebrate (Cooper, 2001). In molluscs, anaesthetic agents can be administered using aqueous solution via immersion of only the foot of the individual. In certain cases cephalopod anaesthetics can require assisted ventilation, with cooled oxygenated anaesthetic being passed either manually or using a pump through the mantle cavity continuously (O'Dor et al., 1990).

Signs of anaesthesia in invertebrates can be similar to signs of death. Overdosage of anaesthesia will result in death. This can be used as a method of euthanasia.

ANAESTHETICS

A number of anaesthetic agents have been used on invertebrates through the years – often leading to sedation of the individual rather than full general anaesthesia, for example smoke has been used as a sedative for bees (Cooper, 2001).

Hypothermia

Hypothermia can facilitate handling, for example 30 min at 4°C for most species will produce immobility, but in Solifuge (camel spiders) it can be fatal (Cooper, 2001). Decreasing temperature can be useful to immobilise insects for non-painful procedures, such as photography or radiography, but should not be employed for any procedure that may cause pain or stress (Cooper, 2006) as it provides no analgesia (Williams, 2002). Change in temperature is known to be a stressor in insects, as are starvation and chemical insult. These factors can affect the immune response (Brey, 1994), leading to an increased weakness to infectious disease, but the effect that hypothermia could have during the length of an anaesthetic has yet to be assessed.

Molluscs can be cooled to 2°C, or even lower in some species, but long periods at low temperatures may cause death, especially in warm water species (Ross and Ross, 1999). In crustaceans cooling to this degree can lead to autotomy of appendages, although Kuriama prawns (*Penaeus japonicus*) were successfully anaesthetised by cooling to 12°C (Patterson, 1993). Cephalopods can be immobilised by decreasing the temperature from 24°C to 3–5°C, but without effective muscle relaxation (Andrews and Tansey, 1981). In sea anemones cooling to 4–10°C can be used as part of a protocol to prevent closing prior to anaesthesia (Moore, 1989).

Cooling should not be used to immobilise invertebrates for painful procedures.

Carbon dioxide

Carbon dioxide gas (CO_2) from a cylinder can be bubbled through water and in emergency situations a product such as Alka Seltzer® can be used to liberate the gas, but there is doubt about the amount of analgesia provided with CO_2 anaesthesia (Cooper, 2001). Carbon dioxide has been used as a sedative in insects in the past, but it may cause fatalities in spiders (Pizzi, 2006). Cockroaches appear to be very tolerant of CO_2 levels and have been reversibly anaesthetised with 100% CO_2, though recovery was prolonged.

CO_2 in aqueous solution using a 50:50 dilution of soda water or an Alka Seltzer® tablet (Bayer, Germany) has been successfully used in leeches (Williams, 2002) as an anaesthetic.

Gaseous anaesthetic agents

The agents most commonly used in veterinary practice for anaesthesia of terrestrial invertebrates are gaseous agents administered via chamber. This method allows minimal handling of the individual animal and relatively rapid anaesthesia and recovery in most species. It provides full anaesthesia and analgesia, although analgesic effects do not continue after recovery has occurred. Isoflurane, the most commonly used of these agents, is known to have a high safety margin in other species, even in debilitated animals. These agents and the method of administration produce a degree of risk of exposure to personnel and, therefore, if possible, scavenging should be carried out.

Halothane gas can be used at up to 10% and isoflurane gas at 3–5% for induction (Williams, 2002). It is important to cover the end of pipes with open weave mesh or gauze to prevent escape, especially in species such as myriapods.

Induction occurs in myriapods within 15–25 min using isoflurane at 5% in 100% oxygen (Chitty, 2006). In decapods gaseous anaesthetic agents are effective in air, by immersion in solution or by injection through the arthroidial membrane (Fig. 19.5), for example 0.5% halothane by volume was used to anaesthetise crayfish (*Astacus astacus*) (Obradovic, 1986) within 15 min (Ingle, 1995). Aquatic gastropods can be reversibly anaesthetised using halothane, enflurane and isoflurane gases (Girdlestone et al., 1989). Invertebrates such as brachiopods, for example *Daphnia magna*, can be anaesthetised by these agents bubbled through water in 100% oxygen. Methoxyflurane has also

been used in this manner (McKenzie et al., 1992), though anaesthesia is temperature-dependent.

Magnesium chloride and magnesium sulphate

Magnesium chloride and magnesium sulphate are some of the oldest agents used with invertebrates (Table 19.2). They are formulated as a saturated solution in distilled water for freshwater animals and are diluted with an equal volume of seawater for marine animals. Iso-osmotic 7.5% w/v magnesium chloride ($MgCl_2.6H_2O$) can also be added to the medium containing the animal until a lack of response to stimulus is noted.

In molluscs, magnesium ions compete with calcium ions required for synaptic transmission thereby leading to immobilisation. These agents do not provide true anaesthesia in many invertebrates and their analgesic properties have yet to be determined (Ross and Ross, 1999). Magnesium chloride works well in cephalopods as the site of action is thought to be the central nervous system, whereas in crustaceans and vertebrates $MgCl_2$ works at the postsynaptic membrane of the nerve–muscle junction (Scimeca, 2006). Many other agents are ineffective in cephalopods, possibly due to the nature of their nerve transmitters and receptors; subtle differences between cephalopod species have also been shown (Messenger et al., 1985).

Alcohol-based anaesthetics (Table 19.3)

In cephalopods the combination of cooling to 5–7°C with ethanol or magnesium anaesthesia allowed for surgery of up to 15 min, though it required assisted ventilation via cooled oxygenated anaesthetic being passed through the mantle cavity continuously. In some cases ethanol anaesthesia can lead to initial hyperactivity in cephalopods that may lead to traumatic damage (O'Dor et al., 1977).

Tricaine methane sulphonate (MS-222) (Table 19.4)

MS-222 is employed widely in fish anaesthesia, but is less predictable when used to anaesthetise aquatic invertebrates. MS-222 has produced varied results in anaesthesia of decapods, though the sand shrimp (*Crangon septemspinosa*) was anaesthetised by 0.5 g/L MS-222 (Foley et al., 1966). MS-222 appears to be ineffective in crayfish (*Astacus astacus*) at lower concentrations (100 mg/L) and has only a mild effect at high dose rates (1000 mg/L) (Obradovic, 1986). MS-222 at 500 mg/L has been shown to take up to 1 h in crustaceans to produce anaesthesia. Although it is approved for aquatic animals not intended for food in many countries, it is not particularly effective in these species (Brown et al., 1996; Gardner, 1997; Oswald, 1977).

Injectable anaesthetics (Table 19.5)

In decapods injectable anaesthesia can be carried by injection into the haemocoel via the arthrodial membrane of a posterior leg or between the ventral integumental plates (see Fig. 19.5). Some work has shown that many agents, other than those listed below, including MS-222, were ineffective or led to limb autotomy when administered by this route (Oswald, 1977).

Miscellaneous agents

A multitude of other agents have been used less frequently in invertebrates. A number of these are summarised in Table 19.6.

Benzocaine, which must first be dissolved in a small volume of acetone, has been suggested for use in aquatic invertebrates via immersion at 100 mg/L in water (Grist, 2001).

Benzocaine, tubocurarine, gallamine, chlorpromazine (Oswald, 1977), carbon dioxide, 2-phenoxyethanol and magnesium chloride (Gardner, 1997) have been shown to be ineffective or detrimental to crustaceans. Xylazine administered at 100 mg/kg led to a number of mortalities and is therefore not recommended at this dose rate (Ferrero and Pressacco, 1982). Agents such as urethane – now known to be carcinogenic – have been used to anaesthetise invertebrates in the past.

> In general, terrestrial invertebrates are anaesthetised using gaseous agents and aquatic invertebrates are anaesthetised using aqueous agents. A notable exception is decapods that are often given injectable anaesthetics.

ANAESTHESIA MONITORING

It is hard to judge the depth of anaesthesia in invertebrates. Lack of movement and righting reflex indicate full anaesthesia, but intermediate stages are harder to judge, for example movement of appendages and muscle tone are often unreliable indicators. In spiders, recovery from anaesthesia is gradual with increasing leg movements and righting attempts over 3–20 min after anaesthesia. Slow ambulation may occur 30–120 min after anaesthesia, but prey should not be offered for 48 h, as live prey may injure a spider still recovering from the after-effects of anaesthesia (Pizzi, 2006). Arboreal species should not be allowed access to climbing material until any incoordination has ceased due to the risk of fatal injury from falling.

In snails, response to pricking of the foot can be an indicator of anaesthetic depth (Cooper, 2001). In myriapods, indication of sufficient anaesthesia involves loss of voluntary movement and loss of righting reflex and withdrawal. Full recovery may take a few hours (Chitty, 2006). In insects, total immobility, absence of righting reflex and apparent lack of awareness to stimuli are often reversible with removal of the anaesthetic agent; therefore, ascertaining whether an insect is dead is often far from easy. Recovery can start with limb twitching and proceed to full recovery within hours. The best method of ascertaining if

Table 19.2: Dose rates for the use of magnesium chloride and magnesium sulphate in a variety of invertebrates

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Annelids			
Polychaetes	Magnesium chloride in water[6,8]	Immersion: concentration of between 7.5 and 8% for relaxation	Some species may display differing sensitivity
Cnidaria			
Calliactis parasitica (sea anemone)	Magnesium chloride[13]	Immersion: 0.3 M until 'sufficient' relaxation	
Echinoderms			
Asterias forbesi (sea star)	Magnesium chloride[1]	Immersion: 8% solution in tap water	
Luidia clathrata and *Astropecten articulatus* (sea stars)	Magnesium chloride[7]	Immersion: $MgCl_2.6H_2O$ 7.5% solution mixed with an equal volume of sea water	
Molluscs			
Bivalves: *Ostrea edulis* (flat oyster)	Magnesium chloride[3]	Immersion 3.5% w/v	Relatively quick induction, 90 min anaesthesia, rapid recovery, minimal stress + mortality
Bivalves: *Pecten fumatus* (commercial scallop)	Magnesium chloride[5]	Immersion 30 g/L	Effective anaesthesia, but excess mortalities
Cephalopods	7.5% magnesium chloride in distilled water mixed with an equal volume of sea water[2, 9,11]	Immersion: 1:1 mixture for surgical anaesthesia and invasive clinical procedures 1:3 or 1:4 for handling and examination, and 1:9 dilution for sedation and shipping	Induction took 13 min and recovery 8 min approximately
Cephalopod, e.g. *Sepioteuthis sepoidea* (Caribbean reef squid)	Using the above preparation of $MgCl_2$ with a 1% solution (by volume) of ethanol added[4]	Immersion	Surgical anaesthesia
Cephalopods, e.g. *Sepioteuthis sepoidea*	Magnesium chloride[4]	Immersion: concentration of 1.5 to 2%	Effective anaesthesia
Cephalopods, e.g. *Sepioteuthis sepoidea*	Magnesium sulphate[4]	Immersion: concentration of 3–4%	Effective anaesthesia
Gastropods	Magnesium chloride[10]	Injected as a 10% solution close to the cerebral ganglia	Quick relaxation for 5–15 min
Gastropod: abalone	Magnesium sulphate[14]	4–22% (wt/vol)	5–8 min
Gastropod: sea hare	Magnesium chloride[12]	0.4 M $MgCl_2$ in sea water with 2 mM Hepes (pH 7.7) injected into haemocoel through the foot	

[1] (Anderson, 1965); [2] (Best and Wells, 1983); [3] (Culloty and Mulcahy, 1992); [4] (Garcia-Franco, 1992); [5] (Heasman et al., 1995); [6] (Lewbart and Riser, 1996); [7] (McCurley and Kier, 1995); [8] (Muller et al., 2003); [9] (O'Dor et al., 1990); [10] (Runham et al., 1965); [11] (Scimeca and Forsythe, 1999); [12] (Smolowitz, 2006); [13] (Westfall et al., 1995); [14] (White et al., 1996)

Table 19.3: Dose rates for the use of alcohol-based agents in a variety of invertebrates

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Annelids			
Lumbricus terrestris (common earthworm) and *Eisenia foetida* (compost worm)	Ethanol in Rushton's Ringer's solution[1]	Immersion: 5% ethanol combined with Ringer's	Immobilised
	Ethanol[7]	Immersion: 5% solution at 23°C	Immobilised
Arthropods			
Crustaceans	Ethanol[12]	Immersion: 5–10% solution	
Crustaceans	Chlorbutanol at varying dose dose rates[12]	Immersion	Can be irritant to humans
Crustaceans	2-phenoxyethanol[12]	Immersion (1.5%)	
Crustaceans	Isobutyl alcohol[12]	Immersion (1.5–7 cm^3/L)	
Crustacean stomatopods, e.g. *Squilla mantis* (mantis shrimp)	Isobutanol[2]	Cardiac injection 100–200 μl/kg, between 11 and 17°C	Reliable anaesthesia
Crustacean: decapods, e.g. *Homarus americanus* (American lobster)	Isobutyl alcohol[3]	Immersion at concentrations of 0.5–14.4 cm^3/L	This caused brief hyperactivity and recovery took 10 min
Cnidaria			
Coelenterates	Ethanol[13]	Immersion	Reduce or eliminate pulsing of the bell Probably also effective in comb jellies
Echinoderms			
Holothuria forskali and *Stichopus badionotus* (sea cucumbers)	Propylene phenoxetol[6,14]	Immersion: 2 ml/L saturated solution in sea water	
Molluscs			
Cephalopods	1.5–2% ethanol in sea water[8]	Immersion	Initial hyperactivity can be traumatic and lead to high mortalities if individuals are left too long in solution
Cephalopods: octopuses and other	Ethanol[5]	Immersion: 20 ml/L	
Cephalopod: *Sepia officinalis* (common Cuttlefish)	Ethanol alone[11]	Immersion: 3% for induction and 1.5% for recirculating maintenance	Surgery
Cephalopod: *Sepioteuthis sepoidea*	Ethanol[4]	Immersion: 1–3%	
Gastropod: (abalone)	2-phenoxyethanol[15]	Immersion: 0.05–0.3 % (vol/vol)	1–2 min
Molluscs	2-phenoxyethanol[9]	Immersion up to 1% solution	Effective in clams
Molluscs	Ethanol (95%)[10]	Immersion: 5–15% solution	2–8 min

[1] (Cooper, 1968); [2] (Ferrero and Pressacco, 1982); [3] (Foley et al., 1966); [4] (Garcia-Franco, 1992); [5] (Grist, 2001); [6] (Hill and Reinschmidt, 1976); [7] (Marks and Cooper, 1977); [8] (O'Dor et al., 1990); [9] (Owen, 1955); [10] (Ross and Ross, 1999); [11] (Scimeca, 2006); [12] (Smaldon and Lee, 1979); [13] (Stoskopf, 2006); [14] (Van den Spiegel and Jangoux, 1987); [15] (White et al., 1996)

Table 19.4: Dose rates for the use of MS-222 in a variety of invertebrates

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Arthropods			
Crustacean: Amphipods, e.g. *Gammarus pullex, Corophium* sp.	MS-222[1,5]	Immersion at 0.5–1.0 g/L	Slower induction, but greater safety margin at lower temperatures
Crustacean: *Crangon septemspinosa* (sand shrimp)	MS-222[4]	Immersion at 0.5 g/L	Anaesthetised
Echinoderms			
Coscinasterias (sea star)	MS-222[6]	Immersion at 10 g/L in sea water	
Molluscs			
Gastropods	MS-222[2]	Immersion 0.3% solution	Reversibly anaesthetised
Mollusc: *Pinctada radiata* (pearl oyster)	MS-222[3]	Immersion at 1 mg/L	Effectively anaesthetised
Cnidaria			
Coelenterates	MS-222[7,8]	Immersion (0.5 g/L)	Immobilised; reduce or eliminate pulsing of the bell This method would probably be effective in comb jellies also

[1] (Ahmad, 1969); [2] (Beeman, 1969); [3] (Ehteshami, 1993); [4] (Foley et al., 1966); [5] (Gamble, 1969); [6] (O'Neill, 1994); [7] (Smaldon and Lee, 1979); [8] (Stoskopf, 2006)

Table 19.5: Dose rates for the use of injectable anaesthetics in a variety of invertebrates

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Arthropods			
Crustacean: Decapods	Alfaxolone-alphadolone (Saffan®)[5*]	30 mg/kg	Anaesthesia
Crustacean: Large lobsters and crabs	Isobutanol[4]	0.1 ml/kg into the abdomen	Produces anaesthesia within 2 min
Crustacean: *Homarus* sp. – lobsters	Isobutanol (100%)[3]	0.2–0.5 μL/10 g into abdominal sinus	Mortality at higher doses
Crustacean: *Pseudocarcinus* sp. – giant crabs	Ketamine HCl[2]	0.025–0.1 mg/kg	Anaesthesia within 15–45 s, can cause rigidity
Crustacean: *Orconectes* sp. – crayfish	Ketamine HCl[1]	90 μg/g IM	Duration 1 h or less
Crustacean *Orconectes* sp.	Lidocaine (lignocaine) HCl[1]	30 μg/g IM injected intrathoracically	Duration 25 min
Crustacean: Decapods	Pentobarbitone[5]	250 mg/kg	Duration 90 min

Continued

Table 19.5: Continued

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Crustacean: *Cancer* sp. – crab – and *Carcinus* sp.– green crab	Procaine HCl[5]	25 mg/kg	Anaesthesia within 20–30 s, duration 2–3 h, short phase (10 s) of excitement
Crustacean: Decapods	Propanidid injection into the haemocoel via the arthrodial membrane of a posterior leg or alternative route[5]	100 mg/kg	Anaesthesia of 60 min
Crustacean: *Cancer* sp. and *Carcinus* sp.	Xylazine HCl[5]	70 mg/kg	Anaesthesia within 5–6 min, duration 45 min
Crustacean: *Pseudocarcinus* sp.	Xylazine HCl[2]	16–22 mg/kg	Anaesthesia within 2–3 min

Key: IM = intramuscular

*Schering Plough Animal Health, Welwyn Garden City, Herts, UK.

[1] (Brown et al., 1996); [2] (Gardner, 1997); [3] (Gilgan and Burns, 1976); [4] (Grist, 2001); [5] (Oswald, 1977)

insects, and most other invertebrates, are truly dead is either to await signs of autolysis or rigor mortis, or to destroy the animal physically so that recovery is impossible. Sick or debilitated insects may succumb to anaesthesia, but generally anaesthesia is relatively safe (Cooper, 2006).

An 8 MHz Doppler ultrasound probe has been used to monitor heart rate under anaesthesia in some species, such as giant land snails (*Achatina* sp.) (Rees Davies et al., 2000) (Fig. 19.16). However, this appears to be ineffective in some other species, for example in millipedes it is difficult to attain good contact on the ventral body (Chitty, 2006).

Judging anaesthetic depth in aquatic invertebrates is even more challenging. Five stages of anaesthesia have been described in leeches, depending on the following factors: attachment to surface, speed and extent of swimming, muscle tone, whether the caudal sucker is functioning, and response to stimulation (Cooper, 2001).

Anaesthesia has been judged to have occurred in the flat oyster (O*strea edulis*) when the valves open but do not close when touched (Culloty and Mulcahy, 1992). In sea anemones depth of anaesthesia is measured by lack of nematocysts (stinging cells) firing, response to feeding or response to tactile stimuli (Moore, 1989).

PERI-ANAESTHETIC SUPPORTIVE CARE

Temperature

Invertebrates should be maintained within their preferred optimum temperature range (POTR) throughout anaesthesia. Although increased temperature can often accelerate anaesthesia and recovery, this can be deleterious in some cases and should only be carried out with care (Cooper, 2001). The POTR can be achieved using heat mats or gloves filled with warm water in terrestrial invertebrates, or by treating aquatic invertebrates in water maintained at a temperature within their POTR.

Fluids

Dehydration is always a threat when the chitinous exoskeleton is breached and insects should always receive fluids before and after surgery, either by mouth via nebulisation or, with caution, intracoelomically (Cooper, 2006). Water or hypotonic saline (0.2–0.5%) can be used. Saline should not be used in phasmids (stick and leaf insects) as their body fluids contain potassium ions, not sodium (Stonehouse, 2003). The exoskeleton of aquatic animals and those with delicate cuticles, such as myriapods (Chitty, 2006), should not be allowed to dry out and, therefore, carrier gas should be humidified.

Oxygen supplementation

During some anaesthetics cephalopods may require assisted ventilation via cooled oxygenated anaesthetic being passed through the mantle cavity continuously (O'Dor et al., 1990). Recovery from anaesthesia can be hastened by pumping oxygen into the anaesthetic chamber or by bubbling it through the water in aquatic situations (Cooper, 2001).

Recovery

Anaesthetised or sedated crustacean individuals should never be returned to tanks housing unsedated tank mates, as these may attack a debilitated individual (Noga et al., 2006). This situation may also occur in other colonies and

Table 19.6: Dose rates for the use of miscellaneous agents in a variety of invertebrates

SPECIES	AGENT	ROUTE/DOSE	COMMENTS
Annelids			
Leeches	Mephenesin (3-*o*-toloxy-1, 2-propanediol)[12]	Immersion	Anaesthesia for grafting research
Arthropods			
Crustacean: *Pseudocarcinus* sp.	Aqui-S (active ingredient Iso-eugenol)[6]	Immersion: 0.125–1.0 ml/L	Induction time 20–70 min
	Chloroform[6]	Immersion: 1.25–2.5 ml/L	Duration 60 min
	Clove oil[6]	Immersion: 0.03–1 ml/L	Induction time >85 min
Crustaceans	Ethane disulphonate[11]	Immersion: 2.5 g/L	
Crustaceans	Methyl pentynol[11]	Immersion: 5 cm^3/L	
Cnidaria			
Adamsia carciniopados (cloak sea anemone) plus various other sea anemone species	Menthol[8]	Crystals ground to coarse powder and scattered on surface of water containing organisms	Lower temperatures slow rate of induction
Echinoderms			
Amphipholis squamata (brittle star)	Menthol[4]	Immersion: 2.5–5% in sterile seawater	
Molluscs			
Gastropod: limpet	Chloral hydrate[4]	Immersion: dilute solution for swimming larvae	
Gastropod: pulmonates	Clove oil (eugenol)[2]	Immersion: 0.6% in water	
Gastropods	Nembutal[9]	Immersion: 0.1%	
Gastropod: abalone	Sodium pentobarbitone[1,10]	Immersion: 60 mg/L of seawater	23°C
Gastropods	Succinyl choline dissolved in sea water at 5 g/L[3]	Immersion: 0.5 mg per 10 g liveweight	Good relaxation
		Injection into space near cerebral ganglia	
Mollusc: *Ostrea edulis* – flat oyster	Benzocaine (must first be dissolved in small volume of acetone)[5]	Immersion: 0.1% solution	
Mollusc: *Pecten fumatus* – commercial scallop	Chloral hydrate[7]	Immersion: 4 g/L	Anaesthesia
Mollusc: *Ostrea edulis*	Chloral hydrate[5]	Immersion: 2–5%	

[1] (Aqualina and Roberts, 2000); [2] (Araujo et al., 1995); [3] (Beeman, 1969); [4] (Costello and Henley, 1971); [5] (Culloty and Mulcahy, 1992); [6] (Gardner, 1997); [7] (Heasman et al., 1995); [8] (Moore, 1989); [9] (Ross and Ross, 1999); [10] (Sharma et al., 2003); [11] (Smaldon and Lee, 1979); [12] (Tettamanti et al., 2003)

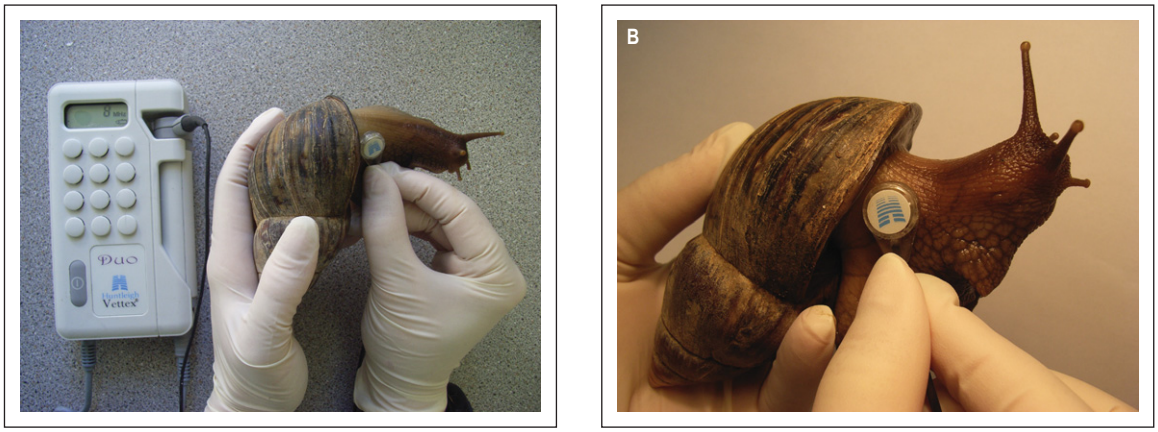


Figure 19.16 • (A) Use of an 8 MHz Doppler ultrasound probe in a giant land snail (*Achatina sp*.). (B) Use of Doppler on giant land snail: close-up.

should be assessed before reintroductions. It is also important that no live food should be left within a tank into which a recently anaesthetised individual is returned, as the animal is unlikely to feed soon after anaesthesia and may, itself, be attacked by other invertebrates supplied as food.

> Do not place recently anaesthetised individuals into an environment with other invertebrates as they may be attacked.

CONCLUSION

Invertebrate anaesthesia is an increasingly vital part of modern veterinary practice and is likely to grow in importance as monitoring methods become more sophisticated. An animal's care should never be compromised simply due to its size, and veterinary clinicians have a duty to provide anaesthesia and analgesia to the best of our abilities to all the many and varied species we treat.

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Appendix

STANDARD NEEDLE SIZES

METRIC	IMPERIAL
13 mm	½″
16 mm	⅝″
19 mm	¾″
25 mm	1″
32 mm	1¼″
38 mm	1½″

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