INTRODUCTION

Reptile anaesthesia is a constantly evolving but frustrating practice. Veterinarians need a reliable, quick acting and safe anaesthetic with consistently reproducible results. Recovery time should be short and uneventful. Many different injectable anaesthetics have been tried in reptiles including ketamine, medetomidine, xylazine, propofol and alfaxalone.

PAIN PERCEPTION

Pain is the conscious perception of nociception, the activation of specialised receptors in response to a noxious stimulus (Heard, 2007). All animals are assumed to have nociceptors and potentially able to perceive pain.

Pain perception in lower vertebrates is likely to be analogous to that of mammals. For this reason alone invasive and painful procedures should always be accompanied by appropriate pain relief and anaesthesia. Although specific doses have not been established in clinical trials, clinicians should attempt to provide lower vertebrates with appropriate analgesia during painful procedures (Machin, 2001). Anaesthetising reptiles is difficult, especially with respect to the monitoring of anaesthetic depth and vital parameters. Methods of support are used less frequently than in domestic species. Studies have shown that provision of pain relief in practice is uncommon. Research regarding pain and its assessment, response to pain relieving agents, and drug pharmacokinetics is needed (Read, 2004).

**Antinociception or “non-responsiveness to noxious stimuli” should not be misinterpreted as analgesia when discussing responses to pain**

K Pasloske

PREPARATION

Reptiles should be kept at their preferred body temperature (PBT) prior to, during, and after an anaesthetic. Most reptiles are not fed daily; consequently it is not difficult to schedule anaesthesia so as to avoid regurgitation of food. As with mammals, it is often beneficial to carry out a complete haematology and biochemistry profile prior to any procedure. However, cost often precludes this from happening in practice. Nonetheless, a complete physical examination should be carried out prior to anaesthesia. Particular attention should be paid to the state of hydration of the patient.

PREMEDICANTS

Butorphanol is commonly used by the author as a premedicant at a dose rate of 0.1 - 1 mg/kg 30 minutes prior to anaesthetic induction. Its use in some species is equivocal. Midazolam is commonly used in crocodiles as a tranquillising agent or as a premedicant
prior to anaesthesia. The author has also used it in marine turtles. Morphine is recommended in some species. One study suggests morphine may be an effective analgesic in iguanas, and buprenorphine was not. In bearded dragons there is evidence that very high doses of morphine were analgesic, but not very high doses of butorphanol. However, the converse appeared to be true in corn snakes.

**INJECTABLE ANAESTHETICS**

**Alfaxalone** (Alfaxan®-CD RTU, Jurox, Australia)
Alfaxalone is a most effective, safe and economical injectable anaesthetic for reptiles.

**Indications for the use of alfaxalone in reptile practice**
- Anaesthetic induction,
- Anaesthetic agent for short procedures
  - Suturing, prolapses, wound debridement, abscess curettage, oesophagotomy tube placement

(See appendix for further information and case reports Carmel, 2002; Simpson, 2004)

**Ketamine and Medetomidine**
Ketamine can be used alone but is more commonly used in combination with an α₂-agonist; usually medetomidine. See Table 1 for dose rates. Ketamine and medetomidine are frequently used in the larger reptiles such as monitors, larger chelonia and crocodiles. The advantage of this anaesthetic regime is its reversibility with atipamezole.

**Table 1. Dosages of anaesthetic drugs used in reptiles**

<table>
<thead>
<tr>
<th>Drug</th>
<th>Route</th>
<th>Dosage</th>
<th>Comment</th>
</tr>
</thead>
<tbody>
<tr>
<td>Alfaxalone</td>
<td>IV, IM, ICo</td>
<td>8 mg/kg IV freshwater turtles (except C. expansa, 4-5 mg/kg IV)</td>
<td>Jugular vein used in C. longicollis, C. expansa</td>
</tr>
<tr>
<td></td>
<td></td>
<td>10 mg/kg ICo - all turtles</td>
<td>Dorsal tail vein or pedal vein in larger short-necked turtles</td>
</tr>
<tr>
<td></td>
<td></td>
<td>5 mg/kg agamids, snakes</td>
<td>Subcaudal tail vein is used in agamids, snakes and blue-tongues</td>
</tr>
<tr>
<td></td>
<td></td>
<td>8 – 10 mg/kg blue-tongues</td>
<td></td>
</tr>
<tr>
<td>Ketamine</td>
<td>IM, ICo</td>
<td>5 - 20 mg/kg turtles (combination)</td>
<td>Used in combination with medetomidine</td>
</tr>
<tr>
<td></td>
<td></td>
<td>3 mg/kg – sea turtles (comb.)</td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td>10 – 15 mg/kg lizards and snakes (comb.)</td>
<td></td>
</tr>
<tr>
<td>Medetomidine</td>
<td>IM, ICo</td>
<td>50 – 100 µg/kg (comb.)</td>
<td>Used in combination with ketamine</td>
</tr>
<tr>
<td></td>
<td></td>
<td>150 – 300 µg /kg (sea turtles)</td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td>150 µg /kg – lizards and snakes</td>
<td></td>
</tr>
<tr>
<td>Atipamezole</td>
<td>IM, IV</td>
<td>500 µg /kg all species</td>
<td>600 µg /kg chelonians</td>
</tr>
<tr>
<td>Isoflurane</td>
<td>Inhalation</td>
<td>2 – 3%, IPPV, O₂ 1L/min +</td>
<td>IPPV for induction 6/min at 4-5%, reduced to 2/minute after 3-5 minutes</td>
</tr>
</tbody>
</table>
Midazolam  IM  1.5-2.0 mg/kg  
Best used with another agent, e.g. ketamine (20-40mg/kg)

Table adapted from Mosley, 2005; Johnson, unpublished.

CROCODILIAN ANAESTHESIA (Olsson & Phalen, 2012a; 2012b)

Recent research (Olsson & Phalen, 2012a; 2012b) has identified effective doses of injectable agents for the anaesthesia of estuarine (Crocodylus porosus) and freshwater crocodiles (C. johnstoni). The study demonstrated that medetomidine induces a safe, repeatable, profound sedative or immobilisation response when administered to juvenile crocodiles over the range of body temperatures that would be encountered by animals in the wild and is readily reversed by atipamezole at these temperatures. Its use as a sole immobilisation agent is acceptable for routine or minor procedures, and has potential as a pre-anaesthetic agent for more invasive procedures even at suboptimal temperatures. These findings in combination with findings from other previous studies make medetomidine the preferred immobilising agent for estuarine crocodiles under most circumstances (Olsson & Phalen, 2012a; 2012b). Midazolam administered in the forelimb of captive estuarine crocodiles ranging from 2 to 3.5 kg provides a predictable onset and duration of sedation enabling physical examination, sample collection and translocation of the animals. Behaviour following recovery appears normal.

Table 2 effective anaesthetic agents for crocodiles (Olsson & Phalen, 2012a; 2012b)

<table>
<thead>
<tr>
<th>Species</th>
<th>Agent</th>
<th>Route</th>
<th>Dose</th>
</tr>
</thead>
<tbody>
<tr>
<td>C. porosus</td>
<td>Medetomidine</td>
<td>IM (forelimb)</td>
<td>1. 0.5mg/kg (3-11kg BW)</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>2. Allometrically scaled dose for adults (150-370kg BW) 0.13-0.17mg/kg</td>
</tr>
<tr>
<td></td>
<td>Midazolam</td>
<td>IM (forelimb)</td>
<td>5mg/kg</td>
</tr>
<tr>
<td>C. johnstoni</td>
<td>Medetomidine</td>
<td>IM (forelimb)</td>
<td>0.75mg/kg (3-11kg BW)</td>
</tr>
</tbody>
</table>

INHALATION ANAESTHESIA

Isoflurane is the most commonly used gaseous anaesthetic in reptiles. Most animals will require intermittent positive pressure ventilation (IPPV) throughout the procedure, except for large varanids (monitors) which will often breathe spontaneously. Induction rate is 6 breaths per minute at 4-5% isoflurane at a flow rate of at least 1L/minute of oxygen. Frequency is reduced to 2 breaths per minute after a surgical plane of anaesthesia is reached. Intubation is recommended except for very small reptiles. Standard uncuffed endotracheal tubes can be used for the larger reptiles, for example, a 2-3mm tube is used in adult carpet and diamond pythons. In the case of smaller reptiles endotracheal tubes can be fashioned from intravenous catheters or various types of medical tubing. Chelonia are very difficult to intubate, requiring topical application of local anaesthetic to relax the glottal opening. Varanids are snake-like in their oral anatomy, having a rostral glottis, which enables easy intubation. Mouth gags are recommended in varanids.

VENTILATION (Longley, 2008)

Reptiles usually require assisted ventilation, either manual or mechanical. The latter is often preferred due to ease of operation and accuracy. The tidal volume in reptiles is larger than that in mammals, but with a lower respiratory rate. Peak airway pressure should be less than
10-15 cm H₂O. There is a tendency to over-inflate the lungs during assisted ventilation. A low pressure should be maintained in reptiles due to their very fragile lungs and air sacs.

**ANAESTHETIC MONITORING**

**Background and basic cardiology**

Visual observation of the heart beat is possible in anaesthetised snakes and some lizards and monitors. Doppler flow monitors are most useful in detecting the heart beat of lizards and turtles. Pedal, palpebral and cloacal reflexes are not reliable indicators of anaesthetic depth in reptiles. Anaesthesia induces loss of motor function in a caudal direction. During recovery the return of function progresses in the opposite direction. Some knowledge of basic reptilian cardiac anatomy and physiology is important in the understanding of how these wonderful animals behave when they are awake and are asleep.

**Basic cardiac anatomy** (Richniw, 2007)

It is easy to imagine that the hearts of reptiles represent an evolutionary stage from fish and amphibians to mammals. However, the success of reptiles in the animal kingdom over millions of years would argue that their cardiovascular systems are equally developed as mammalian or avian systems. The reasons for persisting with a relatively “primitive” heart anatomy are not completely understood. Evolutionary pressures would undoubtedly have resulted in more “advanced” anatomy if it were necessary.

In some respects, the reptilian heart can be viewed as substantially more complex than mammalian four-chambered, sepatate hearts with parallel pulmonary and systemic circulations.

Reptiles exhibit two basic cardiac morphologies or patterns. The first, seen in Chelonia (turtles, tortoises, terrapins) and Squamata (snakes, lizards) comprises a sinus venosus, 2 atria (right and left), a common ventricle that is partially septated ventrally, a single pulmonary artery and 2 aortic arches (left and right). The sinus venosus is separated from the right atrium by a sinoatrial valve; the atria are separated from the ventricle by monocuspid atrioventricular valves, while the ventricle has semilunar valves separating it from the major vessels. The squamate/chelonian heart can be partitioned into several “compartments”. The cavum dorsale, just ventral to the atrioventricular orifice accepts blood from both the atria and acts as the outflow tract for the aortae. It can be divided into the cavum arteriosum, which receives oxygenated blood from the left atrium, and the cavum venosum, which receives blood from the right atrium and from which both the aortic arches originate. Below the cavum venosum lies the cavum ventrale or cavum pulmonale. The cavum venosum and cavum arteriosum are connected by an interventricular canal – basically a large VSD. The second pattern is observed in crocodilia (crocodiles, alligators, caymans) and consists of two atria, two ventricles divided by a complete septum, two aortic arches, and a pulmonary artery. However, the two aortic arches communicate via a foramen, known as foramen Panniza(e), as well as a distal aortic communication (anastomosis). While this anatomy appears somewhat similar to that of mammals, it is important to realize that the left aortic arch arises from the right ventricle along with the pulmonary artery.

At a cellular level, reptilian ventricular cardiomyocytes appear to be substantially different from mammalian myocytes, with a more spindle shape and a lack of T-tubules. Studies of calcium flux in turtle cardiomyocytes suggests that SR calcium release is minimal, and that most calcium involved in excitation-contraction coupling comes from L-type sarcolemmal calcium channels.

**Functional cardiac anatomy and physiology** (Richniw, 2007)

Initial examination of the hearts of reptilia is perplexing to the new-comer. Lack of septation, or origin of a systemic artery from the right ventricle, imply significant admixture of
oxygenated and deoxygenated blood. However, several elegant studies have defined both the pattern of haemic circulation and the functional advantages conferred by such anatomy. Contrast and oxygen saturation studies in the 50s and 60s showed that oxygenated and deoxygenated blood do not generally mix in snakes, but mix to some extent in turtles. These studies (and catheterisation studies) showed that blood from the left atrium was almost exclusively ejected into the systemic circulation, while blood from the right atrium was expelled into the pulmonary artery. Several features of cardiac function were observed that explained the maintenance of discrete circulations.

First, the AV valves are attached medially, so that when they open they form a functional continuation of the ventricular septum, effectively covering the interventricular canal. This allows blood from each atrium to flow into the respective compartments of the cavum dorsale – venosum and arteriosum. Once ventricular systole begins, the blood within the cavum venosum is ejected into the cavum pulmonale and then into the pulmonary artery. Because of the lower pulmonary vascular resistance, all the blood during early systole (first 25%) is ejected exclusively into the PA. As contraction progresses, the cavum pulmonale becomes functionally separated from the cavum venosum so that during the latter portion of systole, blood from the cavum arteriosum is ejected into the aortae. Intracardiac left-to-right shunting is observed in turtles at rest. Approximately 40% of the blood in the heart enters the systemic circulation, while 60% enters the pulmonary circulation, because a small percentage of oxygenated blood is recirculated through the lungs. Similarly, in Crocodilia, the right ventricular content is ejected under normal situations exclusively into the pulmonary artery.

Blood from the left ventricle is ejected into the right aortic arch and then passes through the foramen Panniza into the left aortic arch, despite the origin of the left aortic arch from the right ventricle. Circulatory exclusivity is maintained by relative resistances of the pulmonary and systemic circulations – diastolic pressure in the left aortic arch is sufficiently high that right ventricular blood is ejected into the pulmonary circulation. A unique crocodilian feature of right ventricular outflow is the presence of cog-teeth just below the pulmonic valve. These appear to provide a mechanical resistance to ejection into the pulmonary circulation toward the end of the cardiac cycle, resulting in ejection of blood into the left aorta, and a biphasic right ventricular pressure wave. The depolarisation of the cog-teeth muscles is delayed from the rest of the right ventricle and affected by vagal innervation. Two additional features allow separation of circulations in reptiles: a very long and slow systole, and ventricular “non-compaction”. A long slow systole (often twice as long as diastole) allows for ejection of first the pulmonary blood, and then the systemic blood from the ventricles. The trabeculated ventricular myocardium, allows “trapping” of blood into small compartments. Evolutionarily, there must be a reason for this cardiovascular design. It may lie with the aquatic nature of many reptiles.

Just as mammals have a “dive reflex” which reduces heart rate (by increasing vagal tone), reptiles also respond to submersion with a profound increase in pulmonary vascular resistance, and a decrease in systemic vascular resistance, resulting in a right-to-left shunting of blood, so that pulmonary blood flow is reduced (in some species, it stops), and systemic flow is increased. This is most pronounced in some species of water snakes, but is also seen in turtles and alligators. This is logical when one considers that during prolonged submersion, pulmonary circulation is futile – no oxygenation would be anticipated. During breathing periods in these reptiles, blood flow shunts left to right to allow maximum oxygenation prior to re-submersion. In snakes and turtles, the blood flow is redirected to the aortic arches, while in crocodiles, blood from the right ventricle is ejected into the left aortic arch, while the foramen Panniza contracts to prevent shunting between the arches. Hypoxia induces similar responses.
Recent studies have also documented profound reversible post-prandial cardiac hypertrophy in pythons, thought to be a response to the large increase in metabolic demand that digestion imposes.

**Cardiac location**
Location of the heart varies among lizards from cranial body cavity at the level of the shoulders (skinks, dragons, iguanas) to almost the middle of the body (monitors). In snakes the cardiac position varies depending upon the habits of the snake, with arboreal snakes having the most cranial cardiac position, terrestrial snakes more caudal and aquatic snakes an almost mid-body position.

**Cardiovascular monitoring**

**Heart rate**
Reptilian heart rates vary according to body temperature, size, metabolic state, respiratory state and the absence/presence of painful stimuli.

**Preferred body temperature and its relation to heart rate** (Brattstrom, 1965)
The preferred body temperature (PBT) is the optimal core temperature at which the heart rate should adhere to the following allometric formula:

\[
\text{Heart rate} = 34(W^{-0.25}) \\
W = \text{body weight in kilograms}
\]

**Auscultation**
Heart sounds are usually not able to be detected by auscultation in reptiles.

**Capillary refill**
Capillary refill time is not usually assessed in reptiles due to decreased tissue perfusion and pigmentation of oral mucosa.

**Electrocardiography**
The ECGs of mammals, birds, reptiles and amphibians resemble each other in general form, with clearly defined P, QRS and T components. In reptiles, an SV wave can be observed preceding the P wave. Standard lead positions described for dogs and cats are used for reptiles. Electrodes are placed in the cervical region in lizards with hearts located within the pectoral girdle. In snakes the electrodes are placed two heart lengths cranial and caudal to the heart. In chelonians the electrodes are placed on the skin between the neck and the forelimbs. Reference values do not exist for most species.

**Doppler flow detection**
Doppler flow detection is used for audible monitoring of blood flow. The Doppler flow detector probe should be placed as close as possible to an artery or the heart to detect blood flow. In lizards and snakes the probe can be placed directly over the heart. The lubricated probe can also be turned dorsally and inserted into either the oesophagus or cloaca to detect blood flow. In large reptiles the probe can also be placed over the eye to detect ophthalmic arterial blood flow. In chelonians the probe is placed at the thoracic inlet to detect cardiac flow.

**Arterial blood pressure measurement**
The mean arterial blood pressure is a better indicator of tissue perfusion than systolic or diastolic pressure. The two forms of systemic blood pressure measurement are indirect and direct. Techniques used for measuring indirect BP are oscillometric, automatic and Doppler. Sites used include the legs and tail. Direct arterial BP measurement is used less frequently than indirect due to increased technical difficulty and cost.
Respiratory monitoring

**Blood gas analysis**
Arterial blood analysis requires a “cutdown” technique, often difficult or nigh to impossible in small reptilian patients. Blood sampled by cardiocentesis does not provide a good reflection of systemic (e.g. carotid) arterial values due to cardiac shunting. Reptile blood cells, like avian blood cells, have a high metabolic activity, so that immediate processing is recommended.

**Pulse oximetry**
There is conflicting information as to the usefulness of pulse oximetry as a monitoring technique in reptiles (Diethelm, Mader & Grosenbaugh, 1998; Mosley, Dyson & Smith, 2004). Reptile haemoglobin differs from mammalian haemoglobin. Pulse oximeters are calibrated to measure relative arterial saturation (SpO\textsubscript{2}) in humans, so absolute values will not relate to the SpO\textsubscript{2} of the reptile. However, pulse oximetry can be used on a comparative basis. In most reptiles, an oesophageal probe level with the carotid artery or a rectal probe can be used (Schumacher & Yelen, 2006).

**Capnography** (Schumacher & Yelen, 2006)
End-tidal PCO\textsubscript{2} has become the standard in human anaesthesia for monitoring respiratory performance and estimation of PaCO\textsubscript{2}. However, analysers with high sampling rates (>100ml/minute) are unsuitable for small reptiles. Analysers with low sampling rates (>50ml/minute) are more suitable. Measurement of end-tidal PCO\textsubscript{2} in reptiles is limited by the fact that reptiles can develop cardiac shunts. A report in green iguanas concluded that no correlation exists between end-tidal PCO\textsubscript{2} and arterial PCO\textsubscript{2} values (Hernandez-Divers, Schumacher & Hernandez-Divers, 2004). To a limited extent, trends in measured end-tidal PCO\textsubscript{2} may be useful during anaesthesia. A decrease may suggest airway leaks or obstruction, disconnection of patient from the breathing circuit, or ventilator malfunction (Schumacher & Yelen, 2006).

**Anaesthetic depth** (Heard, 2007)
Depth is determined by the drugs, dosage of anaesthetic agent, species, presence or absence of disease, and physiological status. Increased depth is assumed when muscle tone decreases, reflexes are obtunded and respiratory pattern becomes regular and even.

Palpebral and corneal reflexes are not elicited in snakes and some lizards due to the presence of a spectacle. A surgical plane of anaesthesia is assumed when no muscle movement occurs and physiological changes are minimal or absent. Sudden tachycardia, hypertension, or tachypnoea in response to stimuli indicates inadequate anaesthetic depth or analgesia. The minimum anaesthetic concentration (MAC) is decreased by hypothermia.

In ectotherms (reptiles, amphibians, fish) patients with a body temperature lower than their preferred optimum require a lower maintenance setting. Premedication with opioids and other premedicants also decrease MAC. Has this been reported in reptiles????

Reptiles have longer induction times than birds and mammals because of inefficient respiratory systems and slow circulation times.

During inhalant anaesthetic induction snakes generally relax from head to tail and recover in the opposite direction. The absence of a response to a cloacal pinch is suggestive of an adequate surgical plane of anaesthesia. Similarly the toe pinch response is used in lizards, crocodilians and chelonians. The corneal response is also used in these groups of reptiles.
RECOVERY

Often recovering reptiles will need IPPV with 100% oxygen until the clinician is satisfied that the animal is breathing spontaneously. Reptiles should also be kept at their PBT during recovery. Turtles should have their necks fully extended during recovery. Post anaesthetic fluid therapy may be required depending upon the reptile’s state of hydration and whether fluid loss has taken place. Depending upon the species, size of the patient and health status fluids may be administered intravenously, subcutaneously, intracoelomically or by the intraosseous route. Routinely the author uses a combination of 5% dextrose saline and normal saline in equal parts at the rate of 20-30 mLs/kg body weight once daily. Fluids should always be warmed to body heat.

POSTOPERATIVE PAIN RELIEF

Adequate studies on the pharmacokinetics of pain relieving agents in reptiles are yet to be done. It can be assumed that, as in mammals, similar caution should be shown in reptiles with the use of non-steroidal anti-inflammatory agents (NSAIDs). Recommended doses for commonly used NSAIDs, local anaesthetics and opiates are shown in Table 2.

Table 3. Dosages of some pain-relieving drugs used in reptiles

<table>
<thead>
<tr>
<th>Drug</th>
<th>Route</th>
<th>Duration (hrs)</th>
<th>Dose</th>
</tr>
</thead>
<tbody>
<tr>
<td>Butorphanol</td>
<td>IM</td>
<td>12-24</td>
<td>0.1-1 mg/kg</td>
</tr>
<tr>
<td>Buprenorphine</td>
<td>IM, IV, SC</td>
<td>12-24</td>
<td>0.4-1.0 mg/kg</td>
</tr>
<tr>
<td>Morphine</td>
<td>IM, SC</td>
<td>12</td>
<td>0.4-2.0 mg/kg</td>
</tr>
<tr>
<td>Meloxicam</td>
<td>IM, IV, SC, PO</td>
<td>24</td>
<td>0.1-0.2 mg/kg q 24h</td>
</tr>
<tr>
<td>Carprofen</td>
<td>IM, IV, SC</td>
<td>24</td>
<td>2-4 mg/kg followed by 1-2 mg/kg q24-72h</td>
</tr>
<tr>
<td>Tolfenamic acid</td>
<td>IM, SC</td>
<td>4-12</td>
<td>4 mg/kg q24-72h*</td>
</tr>
<tr>
<td>Bupivacaine</td>
<td>Local</td>
<td>4-12</td>
<td>1-2 mg/kg (&lt;4mg/kg max.)</td>
</tr>
<tr>
<td>Ketoprofen</td>
<td>IM, SC</td>
<td>24</td>
<td>2 mg/kg</td>
</tr>
</tbody>
</table>

Adapted from Moseley (2005); Longley (2008); *Johnson (unpublished)

POINTS TO REMEMBER (after Longley, 2008)

Circulation in reptiles
- Snake, lizard and chelonian hearts are 3-chambered; crocodilians are 4-chambered
  - A pressure differential exists between the chambers, ensuring that oxygenated and preoxygenated blood do not mix
  - Changes in pulmonary resistance will allow shunting of blood either towards (during respiration) or away from (during oxygen starvation, e.g. diving, apnoea, lung disease) the lungs
  - This shunt becomes important when using volatile agents, as uptake of anaesthetic into the systemic circulation is affected
- Reptiles are capable of anaerobic metabolism
- Environmental temperature will affect heart rate and perfusion

Respiration in reptiles
- Chelonia and crocodiles have complete cartilaginous tracheal rings
- Most snakes other than boids only possess a right lung
- Low partial pressure of oxygen is the main stimulus to breathe
- Respiratory pathology results in:
An increase in pulmonary resistance, leading to R→L shunting of blood in the heart and reduced supply to the pulmonary circulation
An increase in anaerobic metabolism to compensate for hypoxia

Comments on opioid use in reptiles (James Wellehan, University of Florida, vin.com consultant)

- “I do end up using hydromorphone in most non-avian reptiles, in combination with meloxicam, tramadol, and local anesthetics. I've been playing around with gabapentin lately, too. I wish I had better evidence for doing any of these.”
- “Studies that really need to be done include examination of reptile genomes for opioid receptors, characterisation of the binding of various opioids to the different receptors, and determination of the effects of stimulating the different receptors.”
- “I am not aware of any solid analgesic data in squamates outside of the toxicofera. The apparently divergent data in snakes and iguanids/agamids illustrates the importance of species differences in opioid use. We really need opioid efficacy data in an individual species. As another example, it takes more etorphine to knock down a Grevy's zebra than it does to knock down a closely related but much larger white rhino.”

SURGERY IN REPTILES

Surgical principles for reptiles do not differ greatly from mammals and birds. Incisions should be made between scales. There is often very little subcutaneous fat. The surgical approach for coeliotomy (opening of the body cavity) in the snake is paramedian, between the lateral and ventral scales, and midline or paramedian in lizards, taking care to avoid the midline vein. A coeliotomy in a reptile should be closed similarly to a mammalian laparotomy, although often the reptile equivalent of the peritoneum is difficult to identify. Consequently, a muscle layer and a skin layer will usually suffice. If in doubt, suture “like to like”. Sutures should be removed 4-6 weeks postoperatively. Many clinicians prefer an evert ing technique for suturing the skin. It is the opinion of the author that good apposition of skin edges is important rather than ensuring eversion takes place, which often creates an “ugly” wound that interferes with skin shedding (ecdysis). In the case of salpingotomy (surgical opening of the oviduct), most practitioners do not suture the oviduct after egg or foetus removal. This technique does not appear to affect fertility or oviposition. Often, depending upon species and size of the snake, multiple skin and oviducal incisions need to be made when surgically removing eggs from dystocic pythons.

For gastrointestinal surgery, just as with mammals, ensure adequate blood supply to remaining bowel in the case of resection. Use similar suture patterns to mammalian practice. Due to the elongated and linear anatomy of snakes the bowel is less “giving”, so resections are often more difficult due to the difficulty in exteriorising the gastrointestinal tract and having less “gut to play with”.

Choice of suture material is similar to birds and mammals. To suture the skin of lizards such as bearded dragons and monitors, snakes and turtles 3/0 to 5/0 PDS or nylon is recommended. The skin of blue tongues is difficult to suture, particularly that of the dorsum and neck. Suture needles with a cutting edge are preferred in this species. Surgical glue is often used to appose small wounds in smaller lizards and snakes.

Maxillofacial injuries in small reptiles
**Fractures of the mandibular symphysis and rostral ramus in lizards**

Often rescued free-living blue-tongued lizards (*Tiliqua* spp.) and bearded dragons (*Pogona* spp.) are presented with traumatic injuries to the mandible.

Many of these fractures can be managed using a simple external fixation technique, which is a modification of a previously described method (Mader & Bennett, 2006).

Sometimes the device may be attached in a conscious reptile, although in most cases anaesthesia is required. Alfaxalone (Alfaxan CD-RTU®) is the recommended anaesthetic in bearded dragons (5mg/kg IV) and in blue-tongues (10mg/kg IV), administered via the ventral tail vein.

**Equipment required**
- 1 paper clip
- Vetbond® tissue glue
- Fixomull® Stretch bandage

**Technique**
- Clean and dry wound
- Attach strips of Fixomull® along ventral aspect of mandibular rami
- Fashion paper clip to conform to normal shape of the mandible
- Apply Vetbond® to Fixomull and attach wire
- Cover with more Fixomull®
- Apply Fixomull® strips ventrally from one ramus to the other if extra stabilization is required. (Figure 4)

Leave the device on for at least 4-6 weeks. Replace as necessary.

**Fractures of the beak of freshwater turtles**

Occasionally turtles will present with facial trauma, more often than not accompanying shell trauma. It is important to assess carapace, plastron and bridge fractures before embarking on treating other injuries (Johnson & Roffey, 2006).

Most freshwater turtles should be anaesthetised to enable a thorough clinical examination of the head and oral cavity. Use alfaxalone at a dose rate of 8mg/kg IV in long necked turtles (*Chelodina longicollis*) and short necked turtles (*Emydura macquarrii*), and 4mg/kg IV in the broad shelled turtle (*Chelodina expansa*). Jugular vein access is easy in *C.longicollis* and *C.expansa*, unlike *E. macquarrii* which as well as having a short neck has a short temper. Larger turtles of this species may be injected via the dorsal tail vein or the metatarsal vein. Smaller turtles may need to be premedicated IM or intracoelomically (ICO) with ketamine (Ketamine 100mg/kg), 5 mg/kg and medetomidine (Domitor®), 50-100 µg/kg after which the jugular vein is much easier to access.

Upper beak and mandibular injuries in turtles may be fixed by tension band wiring (TBW), using acupuncture and hypodermic needles and non-absorbable suture material.

**Technique for upper beak fracture repair** (Johnson, 2006)
- Clean wound
- Appose wound edges and TBW using needles and suture material.
- Replace as necessary

**Maxillary fractures in lizards**

Due to the normal compressed nature of the lizard skull, particularly the blue-tongue, maxillary fractures may be accompanied by significant sinus, eye and calvarium damage.
Common causes of these injuries are dog attack, strikes from lawn mowers and “whipper snippers” and motor vehicles.

Lizards with simple fractures may be rehabilitated. Multiple fractures, significant haemorrhage and fly strike may indicate euthanasia. It is important to make a prognostic decision early as rehabilitation may be a lengthy process.

Treatment involves adequate pain relief using non-steroidal anti-inflammatory drugs such as tolfenamic acid (Tolfedine®), 4mg/kg SC or meloxicam (Metacam®), 0.2 mg/kg SC, antibiotic cover enrofloxacin (Baytril®) 10mg/kg SC q24-48 hrs or ceftazidime (Fortum®) 20mg/kg SC q72 hrs as necessary and assisted alimentation. The latter may involve the use of a crop needle or in more severe cases an oesophagosotomy tube. Care must be taken to avoid further trauma to the animal during the feeding process. A slurry of a readily available invalid diet such as Hills a/d® is recommended. Feed every second day and ensure that the patient has access to adequate cage heating. In order for healing to occur and drugs to be metabolised reptiles must be able to thermoregulate. The preferred body temperature of the blue-tongue is 32-33°C and the bearded dragon 35°C. Repeated anaesthetics may need to be administered to enable proper wound inspection and lavage.

Retained spectacles

Snakes do not have eyelids. The spectacle is contiguous with skin and is shed with every ecdysis (skin slough). Often in pythons and other species of snakes the spectacle will adhere to the new tissue beneath it. Lubrication, gentle debridement and lifting of the dry scale is effective as a treatment.

Subspectacular abscess

Abscess formation may occur between the spectacles. Resection of a “slice of pie” using a size 11 scalpel blade or needle will aid irrigation. Normal saline, often diluted with enrofloxacin, is used to flush out the purulent material. Often multiple flushings are usually required.

Abscess

Resection of the abscess contents and capsule is required. Wounds should be marsupialised or a drain inserted for postoperative flushing, often for several weeks.

Shell injuries in freshwater turtles

Repair techniques

A variety of techniques are used for the repair of shell injuries, including the use of dental composite, orthopaedic wiring, external coaptation using K wires, pins and screws. Wounds are cleaned and debrided with saline or chlorhexidine 0.05% and fracture surfaces ground back to healthy tissue using a Dremel® tool. Alfaxalone (Alfaxan®-CD, Jurox, Australia) is the anaesthetic of choice, used at a dose rate of 8mg/kg body weight in Chelodina longicollis and a lower dose (4-5mg/kg IV) in larger species such as C. expansa, achieving a good surgical plane of anaesthesia for most simple repairs. For longer procedures turtles are intubated and maintained on isoflurane and oxygen. An oesophagotomy tube can be placed in turtles with mouth injuries to ensure adequate nutritional support. Routine antibiosis is used in all cases. Pain relief is provided using butorphanol tartrate (0.1mg/kg SC) and tolfenamic acid at a dose rate of 4mg/kg SC q24-72h. Often turtles will require long term care in a moist environment that excludes submersion. Techniques used may vary from placing turtles in tubs of shallow water to using a custom made device (“turtle steamer”©). The latter consists of one plastic tub placed inside another, with heated water in the bottom
tub and holes drilled in the upper tub in order to provide a more humid but not “wet” environment.

Information in this manuscript mentions the “off-label” use of drugs in reptiles. Veterinarians are advised to make their own enquiries and make a full disclosure to clients when dispensing or using drugs not registered for use in reptiles.

APPENDICES

Alfaxalone (Alfaxan®-CD RTU, Jurox, Australia)

Description
- 10 mg/mL
- Clear, colourless, sterile injectable steroid anaesthetic registered for use in dogs and cats
- Wide therapeutic margin of safety
- Can be used as an induction agent prior to gaseous anaesthesia or as a sole anaesthetic agent for short examinations or surgical procedures

Pharmacology
- Active molecule = alfaxalone (not alfadalone)
- Alfaxalone positively modulates gamma-aminobutyric acid (GABA) receptors in vitro. It has also been reported that positive modulation of GABA(A) receptors in the rat spinal cord can produce antinociception in vivo (Lewbart, 2005).

Action (mammals)
- Surgical anaesthesia produced by alfaxalone provides good relaxation of the abdominal muscles and respiration is usually well maintained.
- A transient decrease in arterial blood pressure occurs in the initial stages of anaesthesia.
- Dogs – transient apnoea
- Recovery uneventful – mammals return to sternal recumbency within 60-80 minutes

Route of administration
- Intravenous
- Intramuscular route of administration
  - No tissue irritation after perivascular or intramuscular injection
- Use of premedicants will reduce the dose of alfaxalone required and may prolong the duration of anaesthesia and the recovery time
  - E.g. acetylpromazine, atropine, methadone, butorphanol tartrate, diazepam and xylazine

Alfaxalone-alfadolone acetate (Saffan® Glaxo, UK)
- This drug is a combination of two steroid compounds that yield excellent results with a good safety margin.
- No longer available in Australia
- Intraperitoneal injection in rats
  - Alfaxalone produced sedative and anaesthetic effects with no antinociception.
  - Alfadolone caused no sedation but it did cause antinociceptive effects equal in magnitude to those produced by Saffan®.
- Conclusions
  - Saffan® produces antinociception in rats when given intraperitoneally by an interaction with spinal GABA(A) receptors.
Antinociception is due to the alfadolone content of the neurosteroid anaesthetic and not the alfaxalone (Nadeson & Goodchild, 2000).

Case reports and personal experiences of alfaxalone use in reptiles
Carmel (2002)
Trials were undertaken using Alfaxan®-CD RTU for intravenous anaesthesia in various species, especially reptiles:

- The use of alfaxalone/alfadolone* in 40 reptiles of 13 species is described. In lizards and chelonians the effect of the drug combination varied from sedation to deep anaesthesia, depending on the dose.
- The effect in snakes varied, from none at all to deep anaesthesia.
- No fatalities occurred and there were no apparent clinical side effects in healthy reptiles.
- Ultra short acting induction agent or injectable anaesthetic for minor or short procedures
- Dose rate of between 2 and 4mg/kg IV gives excellent results, with surgical anaesthesia reached within 2 minutes of administration.
- Intravenous administration is via the tail vein (lizards/snakes), jugular vein (chelonia) or dorsal occipital sinus (crocodiles). The alfaxalone is given slowly IV over 1-2 minutes to effect.
- Only side effects noticed:
  - One snake exhibited muscle fasciculations for several minutes after administration of the alfaxalone. This is reported as a possible side effect in dogs or cats given this drug, especially if not given slowly. Results have been very promising.
- Recovery is quick (10 to 30 minutes) and uneventful.
- Conclusion The use of Alfaxan®-CD RTU for intravenous anaesthesia in reptiles, especially as a short acting anaesthetic to enable intubation & gaseous anaesthesia maintenance, is recommended. A more detailed clinical report on the use of Alfaxan®-CD in reptiles is planned in the future.

* Alfaxalone/alfadolone (Saffan) was also used in this trial in a minority of cases.

Mark Simpson (2004)
- Reports from the manufacturer suggest less cardiovascular side effects and a greater safety margin compared with propofol.
- Dose 2-4mg/kg IV slowly over 120 seconds. Surgical anaesthesia achieved 2 minutes after dosage completed.
- Recovery fast and smooth
- Alfaxan®-CD RTU is highly recommended for IV administration.7

Robert Johnson (2005)
- Routine anaesthetic for chelonia, especially Chelodina longicollis (common eastern long-necked turtle) due to good venous access.
  - Shorter necked or aggressive turtles may require intracoelomic injection with ketamine or alfaxalone. In the case of experienced operators the subcarapacial route or metatarsal and interdigital veins may be used in larger turtles.
  - Agamids (dragons) and pythons are usually premedicated with butorphanol tartrate 30 minutes prior to induction
  - Recovery is smooth – 1-2 hours.
  - Turtles take the longest time to recover, possibly due to the higher dose used
  - Ensure neck is fully extended during recovery
Turtles may occasionally require IPPV (intermittent positive pressure ventilation) with oxygen during recovery.

- **Dose**
  - The intravenous route is the most commonly used except for fractious animals or those with difficult venous access.
  - **Chelonia**
    - *C. longicollis* 8 mg/kg IV
    - *Macrochelodina expansa* (broad-shelled turtle) 4 - 5mg/kg IV
    - Intracoelomic – 10 mg/kg
  - Agamidae, Pythonidae (pythons) Varanidae (monitors) 5mg/kg IV
  - Scincidae (Blue-tongued lizard) 8 -10 mg/kg

Frye (1991) used alfaxalone/alfadolone (Saffan®) combination at a dose rate of 9-18 mg/kg IM.

**REFERENCES**

Anon. Technical notes, Alfaxan®-CD RTU, Jurox P/L, Australia.


ANAESTHESIA AND CRITICAL CARE OF SMALL MAMMALS
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Introduction

Imagine your first week in practice after graduating and you are presented with the following: a rabbit to castrate; a guinea pig with a skin mass; a mammary tumour to remove from a rat, and a leg of a bird to radiograph. If you deal with exotic & unusual pets and wildlife you will be presented with these sorts of challenges. Your first thoughts may be ‘How do I sedate or anaesthetise this animal safely?’ These notes & presentation will come to the rescue and provide you will a practical overview of the concepts of anaesthesia as applied to rabbits, ferrets, guinea pigs, rats and mice. Note that some of the drug dose tables presented are based on Dr Robert Doneley’s anaesthetic notes.

In the limited time we have we will concentrate on providing a logical approach to the concepts of anaesthesia in the species mentioned above. Special attention will be given to any unique attributes of each animal group that impact anaesthesia. You will also be provided with some example anaesthetic protocols for immediate use.

Intended learning outcomes

By the end of the presentation, and after reading and reviewing these accompanying notes, you should have an understanding of the following items as they relate to small mammals:

✓ Anaesthesia principles
✓ Pre-anaesthetic Considerations
✓ Sample anaesthetic protocols
✓ Monitoring anaesthesia
✓ Important issues of anaesthetic recovery in these species
✓ An awareness of where & when to get help (Resources, Formulary)
Anaesthesia: A logical approach

Hopefully you will already have learnt basic anaesthetic principles from other lectures. I will not detail long lists of possible anaesthetic regimes in these notes but I will provide you with protocols that I have found useful for these species in these notes and the lecture presentation. There are many, many variations on anaesthetic protocols that can be used in these pets so I advise you to first become familiar and competent with one or two protocols for each species.

See the individual species section for anaesthetic regimes

Many small mammals, birds and wildlife species can be rapidly induced via isoflurane mask induction. I prefer to avoid using injectable anaesthetic regimes in most small mammal species. Always pre-oxygenate for several minutes before induction. Ferrets, rats & mice are induced extremely quickly with isoflurane – usually within 1 or 2 minutes – with or without premedication. Rabbits become very stressed if mask induction is attempted and will breath hold: it is strongly recommended to premedicate/sedate rabbits before induction.

Anaesthetic monitoring

Monitoring of the anaesthetic by a dedicated nurse is the highest priority.

No matter what gadgets you have, they are no match for a trained assistant

The gold standard is to intubate all patients that are fully anaesthetised. The species particularly difficult to intubate (or require specialised equipment or skills) include rats, mice and guinea pigs. The key to intubation of any exotic pet is to practice as often as you can and be gentle… if you cannot pass the tube after 2 or 3 goes then stop and use a face mask or delay the surgery to another time.

Some anaesthetic monitoring equipment that may be of use includes:

- Rectal thermometer to check core temperature
- Doppler Ultrasound monitor e.g. Parkes brand. These are excellent for monitoring the heart rate and rhythm of exotic pets. This is probably my most frequently used monitoring item
- Pulse oximeter. Note that paediatric or special sensors may be required
- Stethoscope
- Respiratory monitors such as the ApAlert can be of use but may not be sensitive enough for very small patients
- Other patient monitors
- Your eyes & your touch!
• Anaesthetic Record Chart. You can easily modify a dog/cat chart for exotics. . . .
  The main difference is the requirement to record much faster heart and respiratory rates!
• Small Animal Ventilator, e.g. Vetronics, is useful for prolonged surgeries

Heat Loss

Due to the high surface area to body weight of these pets (especially mice & rats) heat loss during anaesthesia is a major concern. Use minimal clipping of fur/hair and sparing use of scrub solutions. Bubble wrap is an excellent, cheap and readily available insulator that can be wrapped around extremities or other parts of the body not undergoing examination or surgery. Probably the best preventer of heat loss in these pets is via the use of convective heat – hot air – pumped around or over the animal. There is now several (expensive) convective heat systems purpose built for use in small animals. Alternatives include using a hair dryer to blow hot air around the patient. Be careful with heat mats in small exotic pets as burns may result if not monitored carefully.

Plan ahead with the surgery to minimise surgical time by having everything ready to go. Being efficient and quick with surgical procedures of small exotic pets is an important factor in increasing patient survival rates and allows faster recovery from surgery.

After surgery it is essential the animal is isolated from any cage mates. This is particularly important after desexing as the animal (especially if male!) may still attempt mating and result in herniation. Note that males of some species, for example guinea pigs, can store viable sperm for several weeks post-castration.

General guidelines for small mammal anaesthesia:

• Use premedicants in most cases. Consider a premedicant with analgesic properties where indicated
• Minimal, if any, fasting before anaesthesia (see individual species notes)
• Minimise heat loss during surgery
• Judicious use of clipping and disinfectants
• Heat loss prevention via heat pads etc
• Don’t fuss… try to ‘get in and get out’ to minimise surgical time
• Pre-oxygenation. This is as simple as placing a face-mask near or over the mouth and nose of patient for 1-5 minutes before inducing anaesthesia.
• Many of these species can be masked down quickly with isoflurane (exception = rabbit)
• Pain relief can be commenced preoperatively
• Intravenous fluids peri-operatively for all patients. Intraosseous fluids, eg. tibial placement, are an alternative to intravenous fluids in very small patients or where intravenous access is difficult.
• Subcutaneous fluids are routinely given in our practice to patients undergoing minor or short procedures.
• Watch the patient closely during the recovery period. Preferably oxygenate until patient able to lift head
• Keep the patient away from cage mates post operatively
• Pain relief post operatively to help prevent self trauma
• Offer food as soon as awake

**Mice & Rats**

• Prior fasting is minimal: 2-3 hours maximum to limit amount of food in mouth

• Premedication is possible with a range of agents (see table below). I use morphine @ 1-5 mg/kg or butorphanol @ 1-5 mg/kg.

• Rats & mice are induced rapidly with isoflurane. Maintain on 1.5 - 3.5%

• Prevent heat loss as discussed above

• Monitor respiration & HR carefully. Use a Doppler for heart rate & rhythm.

• Gentle tissue handling

• Place on heat pad or in heated enclosure for recovery (Beware: overheating)

• Use intradermal skin sutures or tissue glue & isolate from cage mates for a week or so to help prevent dehiscence of surgical wound.

**Ferrets**

Isoflurane mask induction of ferrets is extremely quick, usually within 1-2 minutes and is useful for quick procedures like blood collection or radiographs. Jaw tone will remain for several minutes until a surgical plane of anaesthesia is reached.

Premedicants to consider include morphine (2-5 mg/kg), acepromazine (0.5-1.5 mg/kg), butorphanol - Torbugesic®/Dolorex® - (0.01-0.5 mg/kg) or buprenorphine - Temgesic® (0.01-0.03 mg/kg). The advantage of using an opiate is that not only are you premedicating the animal but an analgesic effect is provided. Diazepam or midazolam (0.25 - 1 mg/kg) can also be used as a premedicant.

**NOTE:** Ferrets are sensitive to butorphanol (0.01-0.05 mg/kg) – use with care (use the lower range dosages) as it can result in deep sedation
Propofol can be used as an intravenous induction agent @ 2-4 mg/kg, given slowly. It can be combined with diazepam or midazolam (0.25-0.5 mg/kg) (also given intravenously). Alfaxan-CD has been used in ferrets, given to effect slow IV.

Ferret sedation using ketamine & medetomidine given intramuscularly
This is a particularly useful combination for minor procedures such as radiology or sedation for blood collection. The ketamine is given at 2.5-5 mg/kg and medetomidine at 0.04-0.08 mg/kg, both given IM (quadriceps or epaxial muscles). I use the lower dose in almost all cases (2.5mg/kg ketamine & 0.04mg/kg medetomidine). The ferret can be intubated for longer procedures. The sedation is partially reversed with atipamezole (same volume). See the excel table at the end of these notes for dose rate calculations using this combination.

Guinea Pigs

Biology:
Weight: 700—1200g depending on breed
Sexual Maturity: Male 3-4 months, Female 2-3 months
Average life span: 5-6 Years (Max 8)

My preferred protocol for guinea pig anaesthesia is to premedicate with morphine @ 2-5 mg/kg sc. Alternative regimes use midazolam, butorphanol or buprenorphine. Mask down with isoflurane after pre-oxygenation. An anaesthetic induction chamber is an alternative.

Rabbits

Rabbits have a very small chest cavity in relation to their body size and are prone to hypoxia during anaesthesia. This, added with the difficulty of intubation, can make rabbit anaesthesia problematic for veterinarians not experienced with this species. As with the other species dealt with in these notes, there are several combinations of drugs that can be used for anaesthesia in rabbits. I will list in point form the most important points to consider when anaesthetising rabbits:

Withhold food for a maximum of 2-3 hours. Although rabbits cannot vomit, taking away all food for a few hours before sedation or anaesthesia with eliminate the small chance that food will obstruct the pharyngeal area.

Always examine the mouth of any small mammal you anaesthetise
- both to check for ingesta, and to assess the teeth

Rabbits are obligate nasal breathers (as are rodents). Thus you can keep a rabbit anaesthetised with a mask placed only over the nostrils and then have free access to work in the mouth (see notes below about intubation)
I advise premedication of all rabbits undergoing general anaesthesia. Attempting a ‘crash induction’ via mask is very stressful and a rapid release of catecholamines results is a markedly compromised patient. Rabbits will breath hold for long periods if a crash induction is attempted.

Like anaesthetic protocols in other species it is best to become familiar and comfortable with one protocol before experimenting with others!

Sedation for minor procedures: Midazolam (Hypnovel) 0.5 - 2mg/kg IM Alfaxalone (Alfaxan-CD®) 0.5-2 mg/kg IV

Pre-oxygenate for at least 2-3 minutes. Induce via facemask with isoflurane. Wrap the rabbit gently in a towel during induction as some have the tendency to make a violent jump during stage 3 of induction! Slowly increase the isoflurane percentage and maintain on 1.5 – 3%

Ideally intubate. This can be quite difficult for the inexperienced. There are several techniques described - see the reference texts. I prefer the ‘blind’ technique: An assistant or the veterinarian holds the rabbit in sternal recumbency and the head is stretched up & back in full extension. A 2-3.5 mm ET tube (Note the small size) is introduced into the oral cavity with one hand whilst palpating the larynx with the other hand. There is usually a gag reflex when the tube reaches the larynx – back the tube off a centimetre or so and time the introduction of the tube to help reduce the risk of laryngospasm; stop attempting intubation if unsuccessful after 2-3 attempts. Be gentle when attempting intubation!

Intravenous fluid therapy during surgery is ideal. Access sites include the cephalic, saphenous and marginal (lateral) ear vein. Intraosseous fluids (tibia in most species) is an alternative in compromised patients.

Example PROTOCOL 1 (for uncompromised patients):

- Butorphanol (Torbugsic®/Dolorex®) 0.5 mg/kg + Ketamine 10-15 mg/kg + medetomidine (Domitor®) 0.1 mg/kg. See Rabbit Anaesthesia chart
- Give Domitor & Ketamine together slow IM. Mix in same syringe.
- The Butorphanol is given SC
- Rabbit should be in sternal recumbency after 5-10 minutes
- The advantage of this method is the ability to reverse the medetomidine completely with atipamezole (Antisedan®)
- Induce surgical plane of anaesthesia via mask induction as described above
Example PROTOCOL 2:
- Fentanyl (Fentanyl or Sublimaze) 50 ug/kg (ceiling dose of 100ug/kg) + glycopyrrolate 0.01 – 0.1mg/kg
- Fentanyl given slow IM
- Glycopyrrolate give SC, anticholinergic helps overcome bradycardic effects of fentanyl. (atropine often ineffective in rabbit as up to 40% have atropinase)
- Protocol developed by Dr David Vella (See references)
- Induce anaesthesia via isoflurane as per protocol 1 or Intravenous agents (various options). Note Fentanyl has short duration of effect: 20 – 40 minutes.

Avoid the use of Zoletil in rabbits: It can cause renal failure.

Small mammal sedation, anaesthesia and analgesia

Small mammal drug dose tables for premedication, sedation, anaesthesia and analgesia are provided below. Please see the accompanying text for notes concerning use of these drugs in small mammals. These are modified from those published in formularies and also from notes by Dr Bob Doneley.

**Premedication dose rates**

<table>
<thead>
<tr>
<th>Drug</th>
<th>Ferrets</th>
<th>Rabbits</th>
<th>Rats</th>
<th>Mice</th>
<th>Guinea Pigs</th>
</tr>
</thead>
<tbody>
<tr>
<td>Acepromazine</td>
<td>0.1-0.3</td>
<td>0.25-1.0</td>
<td>0.5-2.5</td>
<td>0.5-2.5</td>
<td>0.5-1.5</td>
</tr>
<tr>
<td>Diazepam</td>
<td>2</td>
<td>1-5</td>
<td>3-5</td>
<td>3-5</td>
<td>1-5</td>
</tr>
<tr>
<td>Midazolam</td>
<td>1</td>
<td>1-2</td>
<td>1-2</td>
<td>1-2</td>
<td>1-2</td>
</tr>
<tr>
<td>Atropine</td>
<td>0.05</td>
<td>0.8-1.0</td>
<td>0.05</td>
<td>0.05</td>
<td>0.05</td>
</tr>
<tr>
<td>Morphine</td>
<td>0.2-5</td>
<td>2-5</td>
<td>2-5</td>
<td>2-5</td>
<td>2-5</td>
</tr>
</tbody>
</table>

Drug doses for premedication, in mg/kg. Use lower doses when giving IM (cf. SC), or to debilitated, geriatric, obese, or large individuals. Morphine also provides pain relief with premedicant/sedative effects. Higher doses are for pain relief

**Analgesia dose rates**

<table>
<thead>
<tr>
<th>Drug</th>
<th>Ferrets</th>
<th>Rabbits</th>
<th>Rats</th>
<th>Mice</th>
<th>Guinea Pigs</th>
</tr>
</thead>
<tbody>
<tr>
<td>Buprenorphine</td>
<td>0.01-0.05</td>
<td>0.01-0.05</td>
<td>0.01-0.05</td>
<td>0.05-0.1</td>
<td>0.02-0.05</td>
</tr>
<tr>
<td>Butorphanol</td>
<td>0.01-0.4</td>
<td>0.1-0.5</td>
<td>0.5-2</td>
<td>1-2</td>
<td>0.2-2</td>
</tr>
<tr>
<td>Morphine</td>
<td>0.2-4</td>
<td>2-5</td>
<td>0.5-5</td>
<td>2-5</td>
<td>2-5</td>
</tr>
<tr>
<td>Meloxicam</td>
<td>0.2-0.3</td>
<td>0.3-1.5</td>
<td>1</td>
<td>1-5</td>
<td>1-2</td>
</tr>
<tr>
<td>Carprofen</td>
<td>2-5</td>
<td>1.5-4</td>
<td>1-5</td>
<td>5-10</td>
<td>4</td>
</tr>
</tbody>
</table>
Analgesics used in small mammals. Modified from Wenger 2012
Injectable anaesthesia can be used to induce for a gaseous anaesthetic, for short procedures, or when gas anaesthesia is not available. It is difficult to achieve surgical anaesthesia with injectables alone: hint = use inhalation anaesthesia preferably (with premedication)!

Injectable anaesthetic dose rates

<table>
<thead>
<tr>
<th>Drug</th>
<th>Ferret</th>
<th>Rabbit</th>
<th>Rat</th>
<th>Mouse</th>
<th>Guinea Pig</th>
</tr>
</thead>
<tbody>
<tr>
<td>ACP + ketamine</td>
<td>0.05-0.3</td>
<td>0.25-1.0</td>
<td>2.5-5.0</td>
<td>2.5-5.0</td>
<td>0.5-1.0</td>
</tr>
<tr>
<td></td>
<td>10-30</td>
<td>25-40</td>
<td>50-150</td>
<td>50-150</td>
<td>20-50</td>
</tr>
<tr>
<td>Xylazine*** + ketamine</td>
<td>1-2</td>
<td>3-5</td>
<td>3-5</td>
<td>5-10</td>
<td>3-5</td>
</tr>
<tr>
<td></td>
<td>10-30</td>
<td>20-40</td>
<td>40-90</td>
<td>50-200</td>
<td>20-40</td>
</tr>
<tr>
<td>Diazepam + ketamine</td>
<td>1-2</td>
<td>1-5</td>
<td>3-5</td>
<td>3-5</td>
<td>3-5</td>
</tr>
<tr>
<td></td>
<td>10-30</td>
<td>20-40</td>
<td>40-100</td>
<td>40-150</td>
<td>20-50</td>
</tr>
<tr>
<td>Tiletamine-zolazepam</td>
<td>22</td>
<td>DO NOT</td>
<td>50-80</td>
<td>50-80</td>
<td>20-40</td>
</tr>
<tr>
<td></td>
<td></td>
<td>USE</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Propofol</td>
<td>-</td>
<td>5-8 i/v</td>
<td>10 i/v</td>
<td>20-30 i/v</td>
<td>-</td>
</tr>
</tbody>
</table>

Table 3. Injectable anaesthetics in mg/kg i/m, unless otherwise indicated. Use lower doses when giving i/m (compared to s/c), or to debilitated, geriatric, obese, or large individuals.

*** BC note: I avoid the use of Xylazine

Injectable anaesthetic combination for mice

1.5ml of medetomidine (Domitor 1mg/ml)
0.1ml Atropine (0.6 mg/ml)
1ml Ketamine (100 mg/ml)
Dilute in 12.5ml warmed Hartmann’s solution

Dose:
- 0.1ml diluted solution per 10g BW given IP.
- Additional 0.025ml given for maintenance, if required.
- Reversal with 0.15ml per 10g diluted antipamezole (0.1ml = 0.05 mg Antisedan diluted in 5ml warmed Hartmann’s), given IP.

Source: Hardman and Stanley 2001

References and Useful Resources

Association of Exotic Mammal Veterinarians [www.aemv.org](http://www.aemv.org)

Association of Reptilian and Amphibian Veterinarians. [www.arav.org](http://www.arav.org)

The House Rabbit Society [www.rabbit.org](http://www.rabbit.org)


Exotic DVM Journal. An excellent journal dealing with exotic & unusual pets. Published by the Zoological Education Network, PO Box 51749, Lake Worth, Florida 33454-1749, USA. www.exoticdvm.com Some of the general care sections of these notes are based on the ExoticDVM client care sheets.

SAV03 Small Animal Ventilator. www.vetronic.co.uk


AVA Unusual & Exotic Pet Veterinarian Special Interest Group (SIG). A SIG within the AVA. This group deals with all aspects of medicine and surgery of unusual & exotic animals kept as pets. Species involved include rabbits, rodents, guinea pigs, ferrets, fish, native animals as pets and reptiles. There is student membership for this group. Contact the Australian Veterinary Association for details: www.ava.com.au

Emergency and Critical Care of Small Mammals

Small mammals are no exception to the norm – emergencies occur in these species. The concern for the general practitioner is the thought - perhaps panicked – of ‘how to’ stabilize and work-up a critically ill small mammal. These notes and accompanying presentation will outline a logical and practical approach to emergency care small mammals and provide advice on some common emergencies. A list of useful references is provided at the end of the notes.

The bulk of these notes are taken from a presentation titled Emergency and Critical Care of the Guinea Pig and Small Rodents. Much of the discussion is centered on rodents and guinea pigs, however the principles apply equally to the other commonly kept small mammals such as rabbits and ferrets.

Overview

1. Don't Panic

2. Stabilize the patient

3. Act fast by going slow
   - Once stable prioritize your workup

4. Some common emergency conditions

A. Don't Panic

Veterinarians are well trained to deal with a variety of species. This means you do have the knowledge to provide emergency care to species such as guinea pigs, rats and mice. The first mistake often made is to think ‘I don't know how to treat a rat/ mouse/guinea pig’. By applying first principles with some species-specific knowledge as provided in these notes you will have the skills required to tackle many small mammal emergencies.

Real Veterinarians treat more than 10 species!

See the ‘Cheat Sheets’ at the end of these notes for some biological data and clinical pathology reference values. These values are based on published reference values together with Australian pathology laboratory values and are to be used as a guide.
B. Stabilize

Some factors to consider when presented with a small mammal emergency or to hospitalize and stabilize a small mammal:

• **Warmth.** Large surface area to body mass can result in fast heat loss, especially with the critically ill patient. Heat mats, wheat bags, warmed air or warmed fluids are all options. Monitor the core body temperature of the patient.

• **Stress.** Rats, mice and guinea pigs are easily stressed. Ideally have a quiet, dark, separate ward for these patients. Do not house prey species next to predators: placing a cat next to a mouse is not helpful! Perform clinical examination, treatments and diagnostics quietly, gently and efficiently (see 'Act fast by going slow' below).

• **Weight.** Regularly weigh the patient several times per day using accurate digital scales. A small weight gain or loss may be significant and you also require accurate weights for accurate medicating. Kitchen digital scales that weight up to 2kg with 1g accuracy are available for under $40.

• **Oxygen therapy.** This can be provided via a plastic bag placed around the carry cage or over the front of the hospital enclosure. The oxygen tubing is then placed into the bag. A more scientific approach is to use a purpose built ICU enclosure or humidicrib. I have had great success with the Buster ICU enclosure. It is very affordable, folds down to minimal space when not in use and allows accurate adjustment of oxygen percentage. Best of all it is easy to use and my nurses are able to set the unit up within 2 minutes for the critical ill patient. See the references at the end of the notes for details on where to buy this unit.

  o For anaesthetized or moribund animals oxygen therapy of the intubated patient is ideal. Endotracheal tubes are available for exotics such as the Cook® brand. Alternatively you can fashion endotracheal tubes from intravenous giving sets, urinary catheters or even large bore intravenous catheters. Intubating these small mammals is a challenge and is aided via the use of stylets and some way of visualizing the glottis such as a small bore endoscope.

• **Food and Water.** If practical, offer appropriate food and water. Stock up on food items such as Oxbow Critical care, rat and mouse pellets and cans of baby food. Many sick small mammals will still eat and drink voluntarily.
• **Fluid Therapy.** Fluid therapy in these species can be a challenge. Listed below are some points to consider.

**Fluid therapy**

Fluid choice, requirements and delivery method will depend on the species, any laboratory data obtained, condition and skill of the veterinarian. You may choose to give fluids to a critical ill patient by a less than ideal route, such as subcutaneous, rather than stress the animal (and veterinarian!) by spending time failing to access a vein.

1. **Is fluid therapy required?** Determine if fluid therapy is required based on patient history, clinical examination, any available laboratory data and client wishes (financial restraints). If fluid therapy is required go to step 2.

2. **Determine fluid type to administer.** Balanced electrolytes such as Lactated Ringer’s or Hartmann’s solution are good first choices. For patients that may have been anorexic for long periods you may consider a saline and glucose mix. Colloids have been used in small mammals with acute blood loss.

3. **Determine fluid rate.** A maintenance fluid value for the guinea pig is around 100ml/kg/day and 90-100 ml/kg/day for rats and mice (Girling 2003). Basic fluid requirements can be determined as per guidelines for dogs and cats and by assessing laboratory data.

4. **Determine Fluid delivery Method.**

Oral fluid therapy should be used if tolerated. Stomach tubes or crop needles can be used for delivering oral fluids to rats, mice or guinea pigs. 5-10 ml/kg is suggested as a maximum (Girling 2003). Nasogastric or oesophageal tubes can be used in the guinea pig at up to 10ml/kg but regurgitation is a concern (O’Rourke 2003).

Subcutaneous fluids can be given over the dorsum between the shoulder blades in the guinea pig, although this method may be uncomfortable and result in stress in guinea pigs. Rats and mice can be scruffed and subcutaneous fluids given in the scruff of the neck. Hypertonic solutions should not be given subcutaneously.

Intraperitoneal fluid therapy has the advantage of being easy to administer, large volumes can be given at once, and absorption is relatively fast. Disadvantages include the risk of organ puncture, stress, and potential pressure placed on the diaphragm and respiratory system). 1-10ml for rats and mice, and up to 15-20ml for guinea pigs, is given in the lower right quadrant (Girling 2003).
Intravenous fluid therapy can be a challenge for the comprised small mammal patient. The cephalic or saphenous veins can be accessed in some guinea pig. I prefer the cephalic vein using a cut down technique. The jugular vein can be catheterized in an anaesthetized guinea pig, also using a cut down technique. The lateral tail vein may be used in rats and mice to deliver a bolus of 0.25ml (mice) or 0.5ml (rats) of fluids (Girling 2003). Rats have two lateral tail veins, one dorsal tail vein and one ventral tail artery (Brown 2006). The blood volume of the rat is approximately 6.4% of its body weight. The jugular vein is not often used in a clinical situation for fluid therapy of rats or mice.

Intraosseous fluid therapy is the final option. It has the advantage of quick absorption, can be used where venous access is unavailable, and can be used for blood transfusions. Disadvantages include the need for analgesia and sedation/anesthesia, risk of infection and blockage of the needle. The proximal femur or tibial crests are most commonly recommended sites.

**Transfusion confusion?** Whole blood can be transfused in guinea pigs, rats and mice, as blood typing does not appear to be necessary (Girling 2003). Transfuse blood from like species to like species - e.g. from mouse to mouse or rat to rat (Mader 2002)

**C. Act fast by going slow**

Keep the animal alive. Prioritize your workup. Determine what is the most important diagnostic step you need at this point in time: bloods? Radiographs? Ultrasound? Take your time. It is better to keep your patient alive and perform one test at a time, placing the patient back into ICU to stabilize again before performing the next diagnostic step, rather than performing a battery of tests at once on the patient, gaining the diagnosis but having a post mortem to perform.

You may need to sedate or anaesthetize the patient in order to perform diagnostic steps. Balance the risk of sedation/anesthesia against the requirement for obtaining the data.

**Some blood collection sites to consider:** Rat and mouse: Lateral saphenous vein, lateral tail vein in rats, jugular vein. Guinea pig: Lateral saphenous vein, cephalic vein, jugular vein, vena cava (anaesthetized). Remove no more than 10% blood volume at any one time: Maximum 0.14ml mouse, 1.3ml rat and 0.5 -0.7ml/kg for guinea pigs (Bihun and Bihuk 2003, Quesenberry et al. 2003)

**Euthanasia**

Euthanasia of exotic and unusual pets can be a challenge. I recommend the use of two-stage euthanasia (Carmel and Johnson 2008) technique. The patient is first sedated or anaesthetised before the euthanasia solution is given. Rats, mice and guinea pigs can be ‘crash induced’ rapidly using isoflurane. A sedative such as an
opiate or medetomidine may also be given. The euthanasia solution can then be given intra-cardiac once the animal is anaesthetised.

Ferrets will also crash induce within 1-2 minutes with isoflurane mask, alternatively give an injectable sedative such as ketamine, medetomidine or xylazine. Rabbits will usually struggle and become distressed if mask induction alone is used so an injectable agent as mentioned for ferrets should be used. Diazepam or Midazolam @ 1-2 mg/kg IM are also useful agents for sedating a rabbit prior to giving a euthanasia solution.

Summary

Emergency care of small mammals can be challenging. Much of the same basic principles applied to emergency care of other mammals can be applied. By avoiding the ‘fear factor’ of dealing with a species not encountered frequently, practitioners can develop a logical approach to provide competent patient care. A thorough assessment of the patient, careful selection of anaesthetic agents, together adequate monitoring and post-operative care, will result in smooth anaesthesia of small mammals, with few complications.

References


Critical Care for Herbivores. Oxbow Australia, 2 Baraka Court, Mudgeeraba Qld 4213. (07) 5525 1014. www.oxbowaustralia.com