Anaesthesia of the Domestic Rabbit

Clinical Examination

It is important that a full clinical examination of the rabbit is performed prior to any attempts to anaesthetise. Common sense should prevail and harsh sounding chests on auscultation or a history of >12 hours anorexia should immediately sound warning bells. Gastro-intestinal stasis is a potential complication of general anaesthesia and can be life-threatening in rabbits within 24 hours—often due to a *Clostridia* spp. overgrowth and exotoxin release. Remember that rabbits are obligate nasal breathers and that if the nares are pus filled, or there is evidence of dacrocystitis, this will result in respiratory compromise and probably lung pathology.

If a long period of reduced food intake in an obese or over-weight rabbit has occurred—pre-anaesthetic blood tests to check for hepatic lipidosis are advisable.

Pre-anaesthetic Care

Preanaesthetic Management

Rabbits do not need to be starved prior to anaesthetic as they have a tight pre-cardiac sphincter and cannot vomit unless *in extremis*. Indeed it is deleterious to starve rabbits as this will increase the risk of post-anaesthetic GI stasis, hepatic lipidosis and other complications.

Preanaesthetic Medications

*Glycopyrrolate* – This should be used instead of atropine, as 60% of rabbits possess a serum atropinesterase rendering atropine ineffective. Doses of 0.1mg/kg SC may be given. In general, anticholinergics are not often used in routine anaesthetics in rabbits as they tend to increase the viscosity of respiratory secretions and increase the likelihood of airway blockage.

*Acetromazine* – This can be used safely as a premedicant in rabbits, and may provide enough restraint to allow minor radiographic procedures. Doses of 1mg/kg SC are
recommended. Disadvantages include a longer post-anaesthetic recovery phase and therefore return to eating than premedicants which may be part/fully reversed.

*Fentanyl/Fluanisone* – This is the Hypnorm® combination and is often used at 0.5ml/kg bodyweight to induce sedation. It may be used at 0.1ml/kg to provide a light plane of pre-anaesthetic sedation which facilitates gaseous induction. An advantage of this drug (apart from the fact that it is licensed for use in rabbits) is that it is partly reversible (the fentanyl part) with partial opioid agonists such as buprenorphine or butorphanol.

*Medetomidine* – This may be used on its own or combined with ketamine to produce mild sedation right through (in combination with ketamine) to full surgical anaesthesia. Doses of 0.1mg/kg medetomidine SC or IV may be used to provide very light sedation, through to 0.25mg/kg medetomidine with 15mg/kg ketamine to provide anaesthesia.

**General Anaesthesia**

**Injectable Agents**

*Fentanyl/Fluanisone* - This may be used as sedation only on its own at a dose of 0.5 mls/kg intramuscularly (*see data sheets*). This produces sedation and immobilisation for 30-60 minutes, but its analgesic effect due to the opioid derivative fentanyl will persist for some time after. To provide anesthetic depth, fentanyl/fluanisone may be combined with diazepam (0.3 ml Hypnorm® to 2 mg/kg diazepam) intraperitoneally, or intravenously (but in separate syringes as they do not mix), or with midazolam (0.3 ml Hypnorm® to 2 mg/kg midazolam) intramuscularly or intraperitoneally in the same syringe. Alternatively the Hypnorm® may be given IM first and then 15 minutes later the midazolam is given IV into the lateral ear vein. These two combinations provide good analgesia and muscle relaxation with duration of anaesthesia of 20-40 minutes. The fentanyl part may be reversed with buprenorphine or butorphanol given intravenously, or in emergencies the drug naloxone at 0.1 mg/kg intramuscularly or intravenously may be given, but this provides no substitute analgesia.

Fentanyl/fluanisone combinations are well tolerated in most rabbits, but they can produce respiratory depression and hypoxia, which can lead to cardiac arrhythmias and even arrest.
Medetomidine+Ketamine – As mentioned above, medetomidine at 0.25mg/kg combined with 15mg/kg ketamine can provide surgical anaesthesia. Individual rabbits will vary in their response to this combination and the reader should be aware that these drugs are not licensed for use in rabbits. The alpha 2 agonists such as medetomidine produce the same side-effects in rabbits as are observed in cats and dogs with hypotension, hyperglycaemia, cardiac arrhythmias etc are seen. The advantage is that medetomidine may be reversed with atipamazole at 1mg/kg.

Medetomidine+Ketamine+Butorphanol – This combination allows the further reduction of the dosages of medetomidine and ketamine and so minimise their unwanted side-effects. This author has used dosages of 0.1mg/kg medetomidine with 5mg/kg ketamine and 0.5mg/kg butorphanol given SC/IM or by slow IV. This can provide light surgical anaesthesia for 15-20 minutes and may be deepened or prolonged by intubation and volatile gaseous anaesthesia such as isoflurane or sevoflurane. Again, the dose of medetomidine may be reversed with atipamazole at 0.5mg/kg (i.e. the same volume as that of Domitor® given).

Inhalational Anaesthetics

Isoflurane – This is the main gaseous anaesthetic of choice at present, although sevoflurane has some advantages (see below). It has the advantage that it is licensed in rabbits, and respiratory arrest occurs prior to cardiac arrest (unlike the situation with halothane). It is easier to induce and maintain rabbits that have been premedicated with a low dose of fentanyl/fluanisone or an alpha 2 agonist/triple combination. The rabbit may then be induced by face mask as follows:-

In sternal recumbancy a clear face mask is applied to the nose and mouth and 100% oxygen only is supplied for 2 minutes. If regular respiration occurs, the Isotec is turned up to 0.5% isoflurane in 100% oxygen. If regular respiration occurs this is maintained for 2 minutes and then turned up to 1%. The process is continued in 0.5% increments until a surgical plane of anaesthesia is achieved (usually 1.5-2% isoflurane). At this point intubation may be performed or the rabbit maintained on a face mask.

Sevoflurane – This has the advantage that it is less irritant to the mucus membranes and therefore better tolerated by rabbits than isoflurane. It can still be detected though, and
straight masking down with 8% sevoflurane in 100% oxygen as can be performed in dogs, results in breath holding in rabbits. Levels around 3-4% though do not and it may be possible to induce anaesthesia by face mask starting at 3-4% sevoflurane in 100% oxygen. This may then be dropped to 2-2.5% for most minor surgical procedures.

**Intubation**
This may be performed using uncuffed (preferably) ET tubes. A 3kg rabbit will require a 3-3.5mm diameter tube.

Intubation may be attempted by one of two techniques, blind or direct.

*Blind Technique* – The rabbit is placed in sternal recumbancy. The head of the rabbit is grasped one hand the rabbit lifted vertically until its forefeet are just touching the table. The head is kept level and the ET tube is advanced, midline, over the tongue until breathing sounds are detected. The anaesthetist waits until inspiration occurs and quickly advances the tube. Air movement or condensation on the ET tube if transparent will confirm correct placement.

*Direct Technique* – This involves using a 0 Wisconsin blade laryngoscope or an otoscope and placing the rabbit in dorsal recumbancy. The tongue is pulled laterally and the soft palate deflected with the scope to visualise the larynx. A guide wire should be used to enter the larynx, allowing withdrawal of the scope and slotting the ET tube over it and so advancing it into the trachea. The guide wire is then removed. Alternatively a fine endoscope may be inserted through the ET tube as a form of guide wire and direct intubation performed.

In all cases, a small amount of lignocaine spray applied to the tube will aid intubation.

**Anaesthetic Circuits**
Due to their small size, Ayres T pieces and for smaller rabbits (<1kg) Mini Bain or Mapleson C circuits should be used as these minimise dead space and reduce rebreathing.
Anaesthetic Monitoring

Respiration should become regular and even and is often around 30-60bpm. A loss of the pinch reflex in the hind limbs also occurs. Loss of the pinch reflex in the forelimbs in rabbits indicates that the plane of anaesthesia is becoming too deep. Eye placement is not a good indicator of depth of anaesthesia in the rabbit. Protrusion of the eye during deeper planes of anaesthesia is a poor sign and usually precedes cardiac arrest.

Pulse oximetry may be used in rabbits—but many oximeters will not read heart rates above 250 bpm so care should be taken in selection of these. The ear artery may be used, as may the ventral tail artery or a toe artery in larger rabbits.

Side-stream capnography may also be used in rabbits to assess rebreathing and alveolar perfusion in much the same way as for cats and dogs.

A rectal thermometer is useful to monitor for hypothermia—a common sequel in smaller rabbits (normal temperature is around 38.5-40°C). If the body temperature drops below 35°C critical hypothermia has occurred. For this reason the use of radiant heat mats (rather than pressure ones as many smaller rabbits fail to trigger these) or hot air blankets (Bair Huggers) should be used—although care not to create hyperthermia should also be taken!!
Rabbit Emergency Care

Emergency Airway Access and Ventilation (A and B)

Should breathing stop, or hypoxia be detected, then emergency ventilation will be required. This is best achieved by immediate endotracheal intubation.

Intubation may be achieved blindly, with the rabbit in sternal recumbancy. The rabbit’s head is lifted and the ET tube advanced slowly until breathing sounds are heard through the tube. The tube may then be quickly advanced on inspiration. If a transparent tube is used then condensation from the rabbit’s breath when the tube is over the glottis can be seen aiding intubation. If the rabbit has stopped breathing then this technique becomes extremely difficult, therefore a laryngoscope with a Wisconsin 0 paediatric blade can be used to visualise the glottis (Heard 2004). This is best achieved with the rabbit in dorsal recumbancy and the tongue pulled laterally. A guide wire may be inserted through the glottis first and the ET tube threaded over the top. Alternatively a fine endoscope or needlescope may be used as a guide wire instead, threading the ET over the scope prior to intubation. Once through the glottis, the ET tube maybe advanced and the scope retracted easily.

Direct intubation itself may however be difficult in the rabbit owing to the narrow oral cavity and relatively large size of the tongue caudally, or the presence of an obstruction such as a pharyngeal abscess or foreign body. In an emergency therefore it may be necessary to pass a long through-the-needle catheter into the tracheal lumen between two tracheal rings ventrally. A luer adaptor may be attached to allow connection to an anaesthetic circuit for oxygen administration. A tracheostomy may also be performed in the same way as for a cat or dog; the main difference is that some breeds of rabbit, particularly in the doe, have large dew flaps with plentiful subcutaneous fat depots which may make tracheostomy surgery challenging. Otherwise a longitudinal incision is made over the trachea caudal to the larynx, followed by blunt dissection onto the trachea itself. A 180 degree ventral incision in between the tracheal rings 3-4 rings below the larynx is made and an ET tube inserted. This may then be attached to a breathing circuit for ventilation.
If intubation is not possible then either a tight fitting face mask connected to an anaesthetic circuit may be applied, and a high flow rate of oxygen (4-5 litres) or an Ambu bag, to force ventilate the rabbit. Alternatively, moving the rabbit in a see-saw manner may aid ventilation by moving the abdominal viscera backwards and forwards onto the diaphragm and acting as a pump mechanism (Briscoe and Syring 2004). This works on the basis that most of the impetus for inspiration comes from the flattening of the diaphragm rather than the outward movement of the ribcage.

**Cardiovascular Support (C)**

In mammals <10kg, direct cardiac massage by compressing the chest directly over the heart is most effective at increasing thoracic pressure and forcing blood through the arterial vasculature (Henrik 1992). Heart compression rates of 100 beats per minute need to be achieved in rabbits; the technique recommended to maximise cardiovascular output is circumferential chest compression, as is used in human infants, where the chest is compressed over the heart from both sides at once (Costello 2004).

If a cardiac beat is present or a beat is restarted, ECG leads should be applied to discern any dysrhythmias. The type of dysrhythmia reported in rabbits during resuscitation techniques has thus far been different from that reported in cats and dogs. The latter have been associated with electro-mechanical dissociation, whereas in rabbits, profound bradycardia, ventricular asystole and ventricular fibrillation have been reported (Rush and Wingfield 1992). Adrenaline may be used, intratracheally if intubated, or intravenously if no cardiac beat is detected and no ECG trace. In the case of fine ventricular fibrillation, the use of adrenaline has been advocated to convert the electrical activity to coarse ventricular fibrillation which is easier to convert (DeFrancesco 2000) – see table 1 for dosages.

Conversion of coarse fibrillation is based on the use of cardiac massage as described above, or if the clinic has access to defibrillation devices then the use of these externally at 2-10 J/kg (starting at low energies and increasing if no response is achieved) may be performed (Costello 2004). Greater success is achieved with defibrillation devices if three initial countershocks are applied at low energies.
Drug Dosage and route

<table>
<thead>
<tr>
<th>Drug</th>
<th>Dosage and route</th>
</tr>
</thead>
<tbody>
<tr>
<td>Adrenaline (1:1000 = 1mg/ml)</td>
<td>0.2-1mg/kg IV, intratracheally</td>
</tr>
<tr>
<td>Dexamethasone</td>
<td>2 mg/kg IV (use with caution)</td>
</tr>
<tr>
<td>Diazepam</td>
<td>1-3mg/kg IV, IM</td>
</tr>
<tr>
<td>Doxapram</td>
<td>2-5 mg/kg SC, IV, orally q 15mins</td>
</tr>
<tr>
<td>Fluids</td>
<td>100 ml/kg/day maintenance</td>
</tr>
<tr>
<td>Furosemide</td>
<td>1-4 mg/kg IV, SC, IM</td>
</tr>
<tr>
<td>Glycopyrrolate</td>
<td>0.02 mg/kg SC, IM</td>
</tr>
<tr>
<td>Lidocaine</td>
<td>1-2 mg/kg IV; 2-4 mg/kg intratracheally</td>
</tr>
<tr>
<td>Midazolam</td>
<td>0.5-2 mg/kg IV, IM, intranasally</td>
</tr>
</tbody>
</table>

Table 1: Emergency drugs used in rabbits (adapted from Kottwitz and Kelleher [2003])

If severe bradycardia is detected, glycopyrrolate should be used (see table 1) in preference to atropine as 60% of domestic rabbits possess serum atropinesterases making atropine less effective (Okerman 1994).

Lidocaine may be administered intratracheally or intravenously if ventricular arrhythmias such as VPC’s leading to ventricular tachycardia occur (see table 1 for dose). However, lidocaine should not be used in cases of AV block or severe bradycardia (DeFrancesco 2000), which are more commonly seen in rabbits.

Cardiomyopathy and valvular insufficiency with resultant congestive heart failure are also seen in rabbits as is atherosclerosis. Treatment of congestive heart failure initially depends on the use of diuretics such as furosemide at 1-4mg/kg IV, repeated every 4-6 hours as required. ACE inhibitors have been used in rabbits but they are more susceptible to their hypotensive side-effects than cats or dogs. Therefore reduced dosages and regular monitoring of the systolic blood pressure using non-invasive techniques devised for cats is advisable (Girling 2003).
ECG

As mentioned above, should cardiac arrest, or arrhythmias be detected, ECG leads may be applied as for cats and dogs and the trace assessed. See table 2 for some normal values for rabbits.

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Normal Result</th>
<th>Notes</th>
</tr>
</thead>
<tbody>
<tr>
<td>P wave height</td>
<td>0.1-0.15 mV</td>
<td>Deflection is low or negative in lead I and always positive in leads II and III</td>
</tr>
<tr>
<td>P wave duration</td>
<td>0.03-0.04 seconds</td>
<td></td>
</tr>
<tr>
<td>P-R interval</td>
<td>0.05-0.1 seconds</td>
<td></td>
</tr>
<tr>
<td>QRS complex duration</td>
<td>0.015-0.04 seconds</td>
<td></td>
</tr>
<tr>
<td>R wave amplitude</td>
<td>0.03-0.39 mV</td>
<td></td>
</tr>
<tr>
<td>Q-T interval</td>
<td>0.08-0.16 seconds</td>
<td>Change of deflection of the T-wave from positive to negative or vice versa indicates myocardial hypoxia as with cats and dogs.</td>
</tr>
</tbody>
</table>

Table 2: Normal ECG values (lead II) for healthy rabbits (from Kozma et al [1974] and Huston and Quesenberry [2004])

Intensive Care of the Rabbit

Fluid Therapy

If the rabbit is dehydrated or hypovolaemic, shock doses of fluids should be administered. It should be noted that fluids should be avoided post resuscitation in cases of cardiovascular arrest where there is no hypovolaemia/dehydration prior to the arrest as these fluids may decrease myocardial perfusion pressures and diminish overall nutrient delivery through the cerebral and coronary vasculature (Cole et al 2002).

Venous access is relatively straight-forward in the rabbit. The marginal ear vein, jugular vein, cephalic vein and lateral saphenous vein can all be used for IV catheter placement.
Long-term (>2-3days) use of the marginal ear vein may however cause sloughing of the ear tip, although in the author’s experience this is relatively rare with careful venipuncture technique. Use of a topical local anaesthetic cream is recommended prior to placement. In addition, if a sedative such as Hypnорм® (VetaPharma UK Ltd.) is used (a fentanyl/fluanisone combination neuroleptanalgesic drug) peripheral vasodilation is common facilitating ear vein catheter placement. The fentanyl portion of this drug may be reversed using the partial opioid agonists buprenorphine (0.01-0.05 mg/kg) or butorphanol (0.1-0.5 mg/kg). The jugular vein may be difficult to access, particularly in does where there is a pronounced ruff of skin and fat deposits. In addition it forms the main venous drainage for the eye, and thrombus formation may lead to periocular swelling. An Elizabethan collar can be used to prevent the rabbit from chewing or removing the catheter although this will prevent caecotrophy and care should be taken to ensure the rabbit is still managing to eat, otherwise assisted feeding (see below) should be instituted.

Intraosseous catheters may be placed into the proximal femur, in the trochanteric fossa, in a parallel direction to the long axis of the femur. Use a 18-23 gauge, 1-1.5 inch spinal or hypodermic needle. Analgesia should be employed (see table 3) whenever placing an intraosseous catheter as should prophylactic antibiosis such as enrofloxacin (Baytril 2.5% Bayer – licensed for use in rabbits).

Intraosseous and intravenous fluid administration should be accurately titrated using syringe drivers rather than relying on drip sets, as even a small error in fluid administration may be proportionally more significant considering the small size of many rabbits.

**Analgesia**

Analgesia is essential when performing any invasive procedure. In addition, many acute emergency are associated with considerable pain e.g. fractures, intestinal obstructions, pyelonephritis, renal calculi etc. The same care should be taken when using NSAIDs in rabbits as is taken with their usage in cats and dogs, i.e. they should be avoided where renal disease or gastric/intestinal ulceration/perforation is suspected.
### Table 3: Analgesics commonly used in rabbits NB: none are licensed for use in rabbits.

<table>
<thead>
<tr>
<th>Drug</th>
<th>Dose rate</th>
<th>Frequency of dosing</th>
</tr>
</thead>
<tbody>
<tr>
<td>Buprenorphine</td>
<td>0.01 – 0.05mg/kg SC, IM, IV</td>
<td>6-12 hourly</td>
</tr>
<tr>
<td>Butorphanol</td>
<td>0.1 – 0.5 mg/kg SC, IM, IV</td>
<td>2 – 4 hourly</td>
</tr>
<tr>
<td>Carprofen</td>
<td>2.2mg/kg SC, PO</td>
<td>24 hourly</td>
</tr>
<tr>
<td>Ketoprofen</td>
<td>1mg/kg IM, SC</td>
<td>12-24 hourly (&lt;2 days)</td>
</tr>
<tr>
<td>Meloxicam</td>
<td>0.2-0.3mg/kg PO</td>
<td>24-hourly</td>
</tr>
<tr>
<td>Pethidine</td>
<td>10 mg/kg SC, IM</td>
<td>2 – 3 hourly</td>
</tr>
</tbody>
</table>

Other Medications

Many rabbits presented as an acute emergency either have already, or frequently go on to develop gastrointestinal stasis. Providing obstructive causes have been ruled out, the use of prokinetic medications such as cisapride, metoclopramide and ranitidine should be performed (see table 4). Cisapride is becoming increasingly difficult to source. Ranitidine acts to reduce acidity in the stomach which is beneficial as many rabbits with gastrointestinal stasis have punctate ulceration of the stomach lining. In humans it also has prokinetic effects encouraging emptying of the stomach and some increased motility of the small intestine, which seems to be synergised by metoclopramide, this appears clinically to be the case in rabbits in this authors and others opinion.

### Table 4: Gut motility enhancing drugs for rabbits NB: none are licensed for use in rabbits. NB none are licensed for use in rabbits.

<table>
<thead>
<tr>
<th>Drug</th>
<th>Dose rate</th>
<th>Frequency of dosing and notes</th>
</tr>
</thead>
<tbody>
<tr>
<td>Cisapride</td>
<td>0.5mg/kg PO</td>
<td>12 hourly (now difficult to obtain)</td>
</tr>
<tr>
<td>Metoclopramide</td>
<td>0.5mg/kg SC</td>
<td>8-12 hourly</td>
</tr>
<tr>
<td>Ranitidine</td>
<td>2-5 mg/kg PO</td>
<td>12 hourly (in combination with metoclopramide acts to promote motility as well as reducing acidity)</td>
</tr>
</tbody>
</table>
Assisted feeding should be carried out in conjunction with the use of prokinetics. This can start off with easily absorbed essential sugars and amino acids (e.g. Critical Care Formula® Vetark Professional) either syringed into the mouth or delivered via a naso-oesophageal tube. As the rabbit improves clinically this should be stepped up to use proprietary critical feeding formulas such as Science Recovery® (Supreme Petfoods) or Critical Care for Herbivores® (Oxbow Pet Products) or vegetable based baby foods (lactose-free varieties). The disadvantage of the baby foods is that they do not contain fibre and so have little or no prokinetic activity, although they do provide nutrients in an easily digestible form.

The levels of energy required for a debilitated rabbit should approach that calculated for growing to lactating rabbits using the formula $MER = k \times (wt[kg])^{0.75}$ where $k=200$ for growth and 300 for lactation (Carpenter and Kolmstetter 2000). Therefore for debilitation, the following daily energy requirement may be used.

$$MER = 250 \times (wt[kg])^{0.75}$$

To re-populate the intestinal flora, transfaunation of caecotrophs from a healthy rabbit may aid the return of normal bowel function. The use of commercial probiotics designed for rabbits has also been advocated, and reduces the risk of transferring potential parasites and other agents to the debilitated patient.
Anaesthesia of Rodents and Ferrets

Preanaesthetic Care

Preanaesthetic Management

Rodents – These do not need to be starved prior to anaesthesia as none can easily vomit and starvation can enhance post op ileus. Instead a brief (1 hour) period of food withdrawal can be instituted to prevent food being present in the mouth at the time of induction.

Ferrets – Starvation should be performed as ferrets can easily vomit, but it should only be for a short period (generally 2-4 hours) as insulinomas are commonplace in ferrets >1.5 years and the metabolic rate of the ferret is high.

Preanaesthetic Premedications

Atropine

This is used in some species such as guinea pigs and chinchillas where oral secretions are high, and intubation difficult. Doses of 0.05 mg/kg (Mason 1997) have been used subcutaneously 30 minutes before induction. Atropine also acts to prevent excessive bradycardia which often occurs during the induction phase.

Tranquillisers are frequently used to reduce the stress of induction, which plays a large part in the risks of anaesthetising Lagomorphs and Hystricomorphs. These species will breath-hold during gaseous induction, to the point where they become cyanotic.

Acepromazine (ACP)

This can be used at doses of 0.2 mg/kg in ferrets, and 0.5-1 mg/kg in rats, mice, hamsters, chinchillas and guinea pigs (Mason 1997). In general it is a very safe premedicant even in debilitated animals. However, it is advised not to use this in gerbils, as acepromazine reduces the seizure threshold, and many gerbils suffer from hereditary epilepsy. It may be combined with buprenorphine at 0.02mg/kg to provide a smoother induction and analgesia.

Diazepam

This is useful in rodents at doses of 3 mg/kg (Mason 1997) particularly in gerbils.
**Fentanyl/fluanisone**

This combination (known as Hypnorm®) may be used at varying doses as either a premedicant, a sedative, or as part of an injectable full anaesthesia. As a premedicant doses of 0.08 ml/kg for rats, 0.2 ml/kg for guinea pigs (one fifth the recommended sedation doses) can produce sufficient sedation to prevent breath-holding and allow gaseous induction. These doses are given intramuscularly 15-20 minutes before induction. Hypnorm® is an irritant and large doses in one spot may cause post-operative lameness. It can be reversed after the operation with butorphanol at 0.2 mg/kg intravenously, or buprenorphine at 0.05 mg/kg (Mason 1997).

Fluid therapy is also a vitally important pre-anaesthetic consideration and will be mentioned below.

**Induction and Maintenance Agents**

**Ketamine and its Combinations**

**Ferrets**

Ketamine may be used alone for chemical restraint in the ferret at doses of 10-20 mg/kg but as with cats and dogs, the muscle relaxation is poor, and salivation occurs. More often ketamine is combined with other agents such as the alpha-2 drugs, xylazine and medetomidine. In ferrets 10-30 mg/kg ketamine may be used with 1-2 mg/kg xylazine, preferably giving the xylazine 5-10 minutes before the ketamine (Mason 1997). This author has used ketamine at 10mg/kg with medetomidine at 0.15mg/kg and butorphanol at 0.1mg/kg which will provide a light plane of anaesthesia.

**Muridae/Cricetidae**

Ketamine can be used at 90 mg/kg in combination with xylazine at 5 mg/kg intramuscularly or intraperitoneally in rats, with mice and hamsters requiring 100-150 mg/kg of ketamine and 5 mg/kg xylazine (Harkness and Wagner 1989). These combinations provide 30 minutes or so of surgical anaesthesia.

In gerbils the dose of xylazine may be reduced to 2-3 mg/kg as they appear more sensitive to the hypovolaemic effects of the alpha-2 drugs, with ketamine doses at 50 mg/kg.
Ketamine may also be used in combination with medetomidine at doses of 0.5 mg/kg. The advantages of the alpha 2 antagonists are that they produce good analgesia (which the ketamine does not) and that they may be quickly reversed with atipamazole at 0.5-1 mg/kg. Their disadvantages include their severe hypotensive effects, and that once administered any injectable anaesthetic is always more difficult to control than a gaseous one. They also increase diuresis and may exacerbate renal dysfunction.

Hystricomorphs
Ketamine at 40 mg/kg may be used in conjunction with xylazine at 5 mg/kg can be used in guinea-pigs to produce a light plane of anaesthesia. Ketamine at 40 mg/kg may also be used with medetomidine at 0.5 mg/kg for guinea pigs, or ketamine at 30 mg/kg with medetomidine at 0.3 mg/kg for chinchillas (Mason 1997). Reversal with 1 mg/kg atipamazole may be performed. Both of these may be improved after an acepromazine premedication of 0.25 mg/kg.

Alternatively for chinchillas a ketamine (40 mg/kg) and acepromazine (0.5 mg/kg) combination can be used. Induction with these drugs takes 5-10 minutes and typically lasts for 45-60 minutes, but recovery may take 2-5 hours for the non reversible acepromazine combination, hence reducing this drug and using the reversible alpha-2 antagonists may be beneficial, but should be weighed against the greater hypotensive effects of the alpha-2 drugs.

In chinchillas this author prefers a premed dosage of 0.2mg/kg acepromazine with 0.1mg/kg atropine as a premedication SC. This is followed by a SC dosage of 2mg/kg ketamine combined with 0.02mg/kg medetomidine 10-15 minutes later. This combinations allows most dental procedures to be performed as well as radiography etc., although it requires volatile (isoflurane or sevoflurane) gaseous anaesthesia supplementation for surgical planes of anaesthesia.
Fentanyl/Fluanisone (Hypnorm®)

*Muridae*

This may be used as sedation only on its own at a dose of 0.01 ml/30 gm bodyweight in mice and 0.4 ml/kg in rats. Again this produces sedation and immobilisation for 30-60 minutes and may be reversed with buprenorphine or butorphanol as above.

Alternatively it may be combined with diazepam (mice 0.01 ml/30 gm Hypnorm® with 5 mg/kg diazepam intraperitoneally; rats 0.3 ml/kg Hypnorm® with 2.5 mg/kg diazepam intraperitoneally) where the diazepam and Hypnorm® are given in separate syringes as they do not mix, or with midazolam. Midazolam is miscible with Hypnorm® and for rodents the recommendation is that each drug is mixed with an equal volume of sterile water first and then mixed together. Of this stock solution, mice receive 10 ml/kg and rats 2.7 ml/kg as a single intraperitoneal injection. These two combinations provide anaesthesia for a period of 20-40 minutes.

*Hystricomorphs*

This may be used for sedation only on its own at a dose of 1 ml/kg intramuscularly. This may be problematic in guinea pigs as large volumes are required and Hypnorm® is an irritant and may cause lameness when the whole dose is placed in one spot –multiple sites are therefore preferred.

Alternatively it may be combined as above with diazepam (1 ml/kg Hypnorm® and 2.5 mg/kg diazepam) in separate syringes intraperitoneally, or with midazolam by making the stock solution as described in Muridae, and then administering 8 ml/kg of this solution intraperitoneally. Hypnorm® may be reversed with the partial opioid agonists buprenorphine and butorphanol, or with the full antagonist naloxone.

*Isoflurane*

This is now becoming the most widespread used gas for maintenance and indeed induction. Usually a premedication is used with analgesia as it has no analgesic effects post op and it is irritant to the mucus membranes. Its outweighing advantages though are in its safety for the debilitated patient as <0.3% of the gas is metabolism hepatically, the rest merely being exhaled for recovery to occur. Recovery is therefore rapid.
Induction levels vary at 2.5-4% and maintenance usually is 1.5-2.5% assuming adequate analgesia. Breath-holding still occurs, but the practice of supplying 100% oxygen to the patient for 2 minutes prior to anaesthetic administration helps minimise hypoxia, and then gradually introducing the isoflurane, first 0.5% for 2 minutes, then assuming regular breathing, increase to 1% for 2 minutes and so on until anaesthetic levels are reached usually allows a smooth induction.

**Sevoflurane**

This gas has an even lower solubility index than isoflurane and appears to be a safe anaesthetic for most small mammals. They may be masked straight down with this gas in 100% oxygen at a level of 4% sevoflurane (any higher levels still induce breath holding) or premedications may be given and then gaseous induction performed. In this and other author’s opinions it is difficult to maintain a stable level of surgical anaesthesia in the guinea pig where isoflurane appears to be the preferred gaseous anaesthetic.

**Intubation**

This is potentially difficult in most rodents due to their small size. In hystricomorphs, particularly the guinea pig this is further complicated by the palatal ostium, a physical connection between the nasopharynx/soft palate and the larynx making viewing the laryngeal opening via the mouth very difficult.

It may however be possible to intubate even small rodents using an otoscope, a guide wire and cut-down urinary/feeding catheters with an ET portex connector attached.

**Intermittent positive pressure ventilation**

This may be necessary in some individuals who proceed to breath-hold during induction. If intubation is not possible then three options are available:

1. To ensure a tight-fitting face mask and have an Ayres-T piece/Mapleson C/Modified Bain circuit with half litre bag attached which can be used to attempt ventilation.

2. To place a nasopharyngeal tube via the medial meatus of the nose, into the pharyngeal area. Then to supply 4 litres or so of oxygen (to
combat the resistance of the small diameter tubing of 1-2 mm) via this route. This requires a small mammal of small guinea-pig size and upwards.

3 Perform an emergency tracheostomy with a 25/27 gauge needle attached to the oxygen outlet.

**Anaesthetic circuits used**

Most of the small mammals described here are <2 kg in weight. For this reason an Ayres T-piece, a modified Bain or Mapleson C circuit are the best ones to use so removing as much of the dead space as is possible. For larger rabbits, an Ayres T piece is usually sufficient.

**Additional supportive therapy**

**Recumbancy**

During most surgical procedures the method of restraint/recumbancy will be dependent upon the area being operated on. Frequently this necessitates the patient being placed in dorsal recumbancy. This brings some problems to small herbivores as their abdomen to thorax ratio is 2:1 instead of 1:2 as with cats and dogs. More gut-fill therefore means when in dorsal recumbancy more weight on the diaphragm, and so more resistance to inspiration. During lengthy surgical procedures this may lead to apnoea and hypoxia. Placement therefore with the cranial end of the patient elevated above the caudal when in dorsal recumbancy can help in this situation.

**Maintenance of body temperature**

Maintenance of core body temperature is vitally important in all patients to ensure successful recovery from anaesthesia. In small mammals their increased surface area in relation to their volume allows more heat to escape per gram of animal. To help minimise this, the following actions may be taken:

1 Perform minimal surgical scrubbing of the site, and minimal clipping of fur from the area. Do not use surgical spirit as this rapidly cools the skin.
2 Ensure the environmental room temperature is at the warm end of comfortable (>18°C).

3 Place the patient onto either a water circulating heat pad, a Bair Hugger® or use latex gloves/hot water bottles filled with warm water around the patient (making sure that the patient does not directly contact the containers as skin burns may ensue).

4 Administer warmed isotonic fluids subcutaneously/intravenously/intraperitoneally during and prior to surgery.

It is also worth noting that anaesthetic gases have a rapidly cooling effect on the oral and respiratory membranes, and so patients maintained on gaseous anaesthetics will cool down quicker than those on injectable ones, and this will worsen as the length of the anaesthesia increases.

It is worth noting however that hyperthermia may be as bad as hypothermia. Small mammals generally have few or no sweat glands, and so heat cannot be lost via this route. In addition very few actually pant to lose heat, so if over warmed, the core body temperature rises and irreversible hyperthermia will occur. A rectal thermometer is useful to monitor body temperature which should read as follows:

- Rabbits – 37-39.4°C
- Guinea-pigs – 37.2-39.5°C
- Chinchillas – 38-39°C
- Ferrets – 37.8-40°C
- Rats – 38°C
- Mice – 37.5°C
- Gerbils – 37.4-39°C
- Hamsters – 36.2-37.5°C
- Chipmunks – 38°C

**Fluid therapy**

Intra, pre and post-operative fluid therapy is very important in small mammals, even for routine surgery. Again, as with the issue of core body temperature maintenance, the small size, high metabolic rates and relatively large body surface area in relation to
volume of these patients means that they will also dehydrate much faster, gram for gram, than a larger cat or dog. Studies have shown that the provision of maintenance levels of fluids to small mammals during and immediately after routine surgery improved anaesthetic safety levels by as much as 15% in some cases, with higher levels if the surgery was being performed on severely debilitated animals.

It is therefore to be strongly recommended that all small mammal patients receive fluids during and after an anaesthetic whether it be routine or not.

Volumes of fluids administered are taken from the following maintenance fluid levels:

<table>
<thead>
<tr>
<th>Species</th>
<th>Maintenance fluid levels</th>
</tr>
</thead>
<tbody>
<tr>
<td>Rabbits</td>
<td>80-100 ml/kg/day</td>
</tr>
<tr>
<td>Ferrets</td>
<td>75-100 ml/kg/day</td>
</tr>
<tr>
<td>Rodents</td>
<td>90-100 ml/kg/day</td>
</tr>
<tr>
<td>Chinchillas</td>
<td>100 ml/kg/day</td>
</tr>
<tr>
<td>Guinea-pigs</td>
<td>100 ml/kg/day</td>
</tr>
<tr>
<td>Chipmunks</td>
<td>100 ml/kg/day</td>
</tr>
</tbody>
</table>

The fluids which may be used for this therapy will obviously depend on the health status of the small mammal patient, but a commonly used fluid for routine surgery is Aqupharm® No 18, 0.18% saline, 4% glucose (isotonic fluids). If dehydration is suspected or diagnosed then Aqupharm® No 3, 0.9% saline, 5% glucose should be used to replace the deficit (as for cats and dogs 1% dehydration = 10 ml/kg/day replacement levels in addition to maintenance requirements). Fluid deficits, as with cats and dogs should be replaced gradually. A rough guide is as follows:

- **Day 1** Replace 50% of fluid deficit + maintenance fluids
- **Day 2** Replace 50% of fluid deficit + maintenance fluids
- **Day 3** Maintenance fluids.
Colloidal fluids should be contemplated if intravenous access can be achieved when haemorrhage occurs, and the use of lactated Ringer’s solution for conditions where potassium loss and metabolic acidosis may occur (e.g. renal disease, chronic diarrhoea).

The routes by which these fluids may be administered will depend on the species and the degree of dehydration. If the fluids are required for routine post/intra-operative requirements with minimal/no dehydration, then the subcutaneous route is satisfactory. The scruff region or the lateral thoracic wall may be utilised in any small mammal patient. For more severe dehydration the intraperitoneal route or intravenous routes (in the larger patients) or intraosseous routes are required.

Intraperitoneal injections require the patient to be placed in dorsal recumbancy with the head tilted downwards, so causing the abdominal viscera to move cranially and away from the injection site which is in the right lower quadrant of the ventral abdominal wall. The needle is angled between 20-40° in a cranial direction and advanced until it just pops through the peritoneum.

In the larger species such as rabbits and ferrets and even guinea-pigs the intravenous route may be used. The lateral saphenous or cephalic veins may be used in guinea-pigs and ferrets, again preferably with 25-27 gauge butterfly catheters. In guinea-pigs, chinchillas and ferrets the jugular veins may also be used in a cut-down procedure which necessitates some form of sedation. In rats and mice the lateral tail veins may be used to administer small volumes of fluid. A 27 gauge needle/butterfly catheter will be required as well as warming the tail to improve vessel dilation.

For those species which have small or reduced peripheral circulation such as gerbils, hamsters or shocked patients, the intraosseous route can be used. In the majority of species the proximal femur is the site of choice. Because of the confining nature of the bone marrow cavity, only small boluses of fluid can be administered, and so an infusion device is important, such as a syringe driver. Asepsis must be strongly adhered to in the case of intraosseous catheters as osteomyelitis can easily ensue in debilitated patients.
Monitoring anaesthesia
This becomes more and more difficult as the size of the patient decreases. No one factor (as with cats and dogs) will allow you to assess anaesthetic depth. Eye position should not be used to assess depth of anaesthesia in small mammals. Instead a useful method is to assess depth by the response to noxious stimuli such as pain.

Initially though the first reflex lost is usually the righting reflex. The next reflex to be lost for example in rabbits and guinea-pigs is the swallow reflex, however this may be difficult to assess. Palpebral reflexes are generally lost early on in the course of anaesthetic, but rabbits may retain this reflex until well into the deeper planes of surgical anaesthesia. The palpebral reflex is also altered by the anaesthetic agent chosen, with most inhalant gaseous anaesthetics causing loss of the reflex early on, but it is maintained with ketamine.

The pedal withdrawal reflex is useful in small mammals, with the leg being extended and the toe firmly pinched. Loss of this reflex suggests surgical planes of anaesthesia, but rabbits again will retain the pedal reflex in the forelimbs until much deeper (and often dangerously deep!) planes of anaesthesia are reached. Other pain stimuli such as the ear pinch in the guinea-pig and rabbit are useful, loss of this indicates a surgical plane, as does the loss of the tail pinch reflex in rats and mice.

Monitoring of the heart and circulation may be performed in a conventional manner with stethoscope and femoral pulse evaluation, or in the larger species using an oesophageal stethoscope. As with cats and dogs the detection of increases in the respiratory and heart rates can be used to indicate lightening of the plane of anaesthesia. More sophisticated techniques may also be used with pulse oximetry to monitor heart rate and haemoglobin saturation. As with cats and dogs, the aim is to achieve 100% saturation and levels below 92% would indicate dangerous hypoxaemia and the initiation of assisted ventilation. The ear artery is useful for this in rabbits using the clip-on probe, conversely the linear probes may be used successfully on the ventral aspect of the tail in most species (lateral tail in rats and mice). Other forms of cardiac monitoring include ECG trace, which is frequently adapted to minimise trauma from the alligator forcep attachments by substituting these for fine needle probes, or by blunting the alligator teeth.
An extremely useful monitoring device is the Doppler probe which can detect blood flow in the smallest of vessels up to the heart itself.

Respiratory monitors may also be used if the patient is intubated and many pulse oximeters have outlets for these allowing assessment of respiratory rates. It is important to obtain an oximeter that can read high heart rates (e.g. VetOx by Heska). Even so—many small rodents have heart rates too fast for any current affordable pulse oximeter.
**Recovery and analgesia**

Recovery from anaesthesia is improved with the use of suitable reversal agents if available. Examples include the use of atipamazole after medetomidine anaesthesia/sedation, and the use of naloxone, butorphanol or buprenorphine after opioid /Hypnorm® anaesthesia/sedation.

Most small mammals will benefit from a quiet darkened and warm recovery area allowing a controlled recovery. Subsequent fluid administration the same day is frequently beneficial as many of these creatures will not be eating as normal for the first 12-24 hours.

Analgesia is vitally important in the quick and smooth recovery process. Return to normal activity such as grooming, eating, drinking etc has been shown to be considerably shortened following adequate analgesia.

Analgesics frequently used in small mammals include the following table:

<table>
<thead>
<tr>
<th>Drug</th>
<th>Ferret</th>
<th>Rabbit</th>
<th>Rodent</th>
<th>Chinchilla (pig)</th>
<th>Guinea-pig</th>
<th>Chipmunk</th>
</tr>
</thead>
<tbody>
<tr>
<td>Butorphanol</td>
<td>0.3 q4h</td>
<td>0.3 q4h</td>
<td>1-5 q4h</td>
<td>2 q4h</td>
<td>2 q4h</td>
<td>1-5 q4h</td>
</tr>
<tr>
<td>Buprenorphine</td>
<td>0.02 q8h</td>
<td>0.03 q8h</td>
<td>0.07 q8h</td>
<td>0.05 q8h</td>
<td>0.05 q8h</td>
<td>0.05 q8h</td>
</tr>
<tr>
<td>Carprofen</td>
<td>4 q24h</td>
<td>4 q24h</td>
<td>5 q24h</td>
<td>5 q24h</td>
<td>5 q24h</td>
<td>4 q24h</td>
</tr>
<tr>
<td>Flunixin</td>
<td>1 q24h</td>
<td>1.5 q24h</td>
<td>2.5 q24h</td>
<td>2.5 q24h</td>
<td>2.5 q24h</td>
<td>2.5 q24h</td>
</tr>
<tr>
<td>Meloxicam</td>
<td>0.2 q24h</td>
<td>0.2 q24h</td>
<td>0.2 q24h</td>
<td>0.2 q24h</td>
<td>0.2 q24h</td>
<td>0.2 q24h</td>
</tr>
</tbody>
</table>

Values are in milligrams per kilogram bodyweight.
As with cats and dogs and indeed humans, the administration of analgesia prior to the onset of pain makes for the most effective control of pain. If this is to be considered, then COX-2 inhibitors are preferred, specifically meloxicam or carprofen.

References


