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Small Mammal Essentials

Mini Series

Session One: Critical care, analgesia and anaesthesia of small mammals

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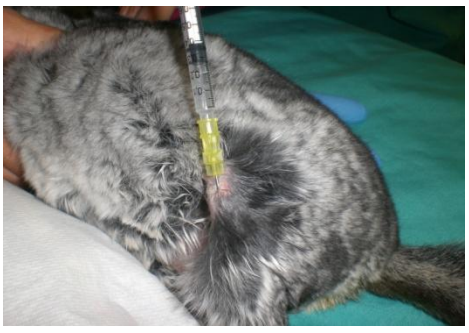

Session 1: Critical care, analgesia and anaesthesia of ferrets and rodents

Small mammals that are commonly seen in practice typically comprise of prey species such as myomorphic and hystrichomorphic rodents and lagomorphs. Prey species have a preservation response and so hide disease until they can no longer pretend to be 'healthy'. They also do not exhibit florid displays of pain or behaviour and so subtle changes in the animals demeanour or behaviour will be missed by most owners.

As a result many can be critically ill by the time they present to the veterinary clinic. These animals will benefit from a period of intensive and supportive care whilst they are further evaluated by the clinical team. This can include detailed (and if possible remote) observation of the patient. Ideally house prey species away from predator species in a dedicated ward. This reduces the stress on these species and allowing them to exhibit normal behaviour. Cats, dogs, ferrets, raptors and snakes should all be housed elsewhere. Ferrets typically spend over 18 hours a day sleeping intermixed with short bursts of activity, so they can be housed anywhere as they typically spend most of their time asleep. This makes them good inpatients.

Prey species are social animals and need companions. Isolating a patient undergoing treatment may well lead to anxiety and stress on that individual. Many owners and their pets may appreciate a 'friendly' companion being present during the hospitalisation period. This may limit the assessment of faecal and urine output and food and water intake, if housed together, so isolation may be indicated for clinical reasons or during the perioperative period, but daily mixing of the individuals is possible. In our clinic animals are typically housed alone with joint periods of access together in the ward until the patient is stable and less observation is required. This will have the additional benefit of reducing the likelihood of upsetting any social hierarchy at home, avoiding conflict between animals.

Prey species should also be able to hide away from their human carers. A quiet ward is ideal with a window or CCTV to allow remove observation, but in many cases wards can be busy due to the cleaning, feeding and treatment of patients. Cardboard boxes suit the requirements for a disposable hide very well and should be included in every cage. Smaller rodents will also appreciate lots of bedding, which can be useful to allow them to hide away. This also provides increased insulation of that animal, allowing more energy to be directed towards healing and supporting body condition as opposed to maintaining body temperature. Ferrets do appreciate areas to hide and are most happy with towels within which they can hide and curl up.

The thermal tolerance of these species can be poor. This is due to their high metabolic rate and large surface to volume ratio. Patients can suffer hypothermia when ill, during the winter or in the perianaesthetic period. However supplemental heating or a warm ward (during the summer if not air conditioned) can lead to hyperthermia. Ward temperatures should ideally not exceed 22 degrees.
Critically ill animals may require supplemental heating. Typically incubators are used elevating the entire temperature to the mid twenties and typically focal heat sources are provided should they be required. This allows the animal to regulate its own temperature with a reduced risk of overheating.

Bodyweight is an important parameter to measure. Small mammals can easily have their weight over or underestimated which can have a major effect on therapeutics. Recording the weight of a patient once or twice a day and then comparing this to previous records (from a consultation for example or from species records if appropriate) will allow the clinician to determine if the patient is sustaining itself or if supportive nutrition is indicated.

Scales used should reflect the size of the patient and in small animals weight records to the nearest gram are required (myomorphic rodents and degus) in others weighing to 10 grams is sufficient (hystricomorphic rodents and ferrets). Postage weighing scales are adequate and plastic boxes can be used to contain an animal whilst it is being weighed.

Small mammals have a high metabolic rate and so food and water should be provided. This also needs to be in a familiar fashion. Guinea Pigs for example may be used to drinking from a bowl or a fountain and the right method should be offered. Many species prefer to drink from a bowl but run the risk of drowning or hypothermia should they fall in if weakened.

The diet should not be changed as this can lead to gastrointestinal upset or reduced intake which is of concern in all species. The hystricomorphic rodents pose an additional problem regarding gastrointestinal stasis, which can lead to dehydration of the bowel contents, impaction and gas formation, which can be intensely painful.

Getting the owner to bring some familiar food items and treats can stimulate voluntary feeding when the animal is ill or after anaesthesia. Many owners will be happy to go away and come back with a suitable platter of food for their pet during the stay at the vets. You also get the opportunity to evaluate the diet the owner perceives as important for their pet. This may well prompt further discussions about improving the diet once their pet has returned home. It is wise to have suitable stocks available as a backup.

Baseline parameters should be taken for every inpatient and compared to previous records for that individual and species ranges. However some of the reference ranges are wide and comparing the values of that patient (lets suggest under anaesthesia or in the immediate post operative period) to records obtained prior to procedures or yesterday can be very useful. There is a huge variation in patient size and physiological state. Parameters should be taken either remotely, or at times of reduced anxiety to increase the reliability.

Physiological data for small mammal species

Mammal	weight range (g)	rectal temperature (^{o}C)	approximate pulse rate/minute	approximate respiratory rate/minute
chipmunk	100-250	38	200	100
chinchilla	400-600	35.4-38	100	45-65
Ferret	600 – 1400	37.8 – 40	200 – 400	33 -36
guinea pig	500-1100	38	230-380	70-100
hamster	85-120	37-38	280-500	35-120
gerbil	45-130	39	260-600	90
mouse	20-60	37.4	300-700	150-200
gerbil	50-90	39	260-600	70-120
rat	250-400	38	300-500	80-100

Supportive care measures can be kept fairly basic. These include providing warmth, oxygenation, fluid therapy, nutritional support (urgent due to metabolic rate and the risk of gastrointestinal slowdown) and attention should be paid to analgesia. Specific diagnostics, emergency procedures or treatments may be indicated in addition to these measures based on clinical presentation.

Providing warmth is important to avoid the risk of hypothermia in ill patients or those in the perianaesthetic period. Some sources may cool over time and run the risk of chilling a patient, others provide a continuous stable heat source and other methods can be used to conserve endogenous body heat generated. Each source has to be considered on its own merits and some can be damaged by a conscious patient (for example a water recirculating heat mat can be chewed).

Ways to reduce the risk of hypothermia

Increasing insulation and decreasing heat loss.	**Providing a continuous heat source.**	**Heat sources that may cool if not monitored or reheated.**
Wrapping extremities in bubble wrap**.**	Warm parenteral fluids in infant bottle warmer.	Hot hands (gloves with warm water).
Warm scrubbing fluid prior to use in infant bottle warmer and avoid or reduce alcohol.	Theatre lighting used as a radiant heat source (watch dessication).	Warm parenteral fluids in the microwave or hot water.
Portex thermovent or warming the oxygen flow to the patients.	Raise room temperature with a heater on a thermostat.	Microwavable bean bag**.**
Minimise spirit use.	Electric heat mats.	Snuggle safe.
Space blanket.	Bair hugger.	Hot water bottle.
Wrapping the animal in a towel.	Water recirculating heat mats.	**ABOVE ALL MONITOR THE RECTAL TEMPERATURE CONTINOUSLY.**
Using plastic drapes as opposed to cloth or paper drapes (no wicking of fluids and better insulation).	Hair dryer (can combine with a bubble wrap cover to create a warmed area. Holes for surgery can be made).	
Reduce surgical time.	Heat lamps.	
Minimise clipping area.	Hot Dog.	
Wrapping extremities in silver foil.	Heated theatre table.	
Switch off air conditioning.	Incubators (in recovery). Plant propagators can also be used.	

Temperature can be monitored using rectal thermometers, which have the disadvantage of being lost under surgical draping. Digital units with a probe can allow remote continuous recording, provided the probe is not dislodged. If the temperature is reducing or increasing remedial action can be taken at an early stage. Alternatively oesophageal temperature can be recorded. The temperature of the patients surface and the environment can also be monitored remotely using a non-contact infrared digital thermometer.

Oxygenation is an important part of critical care. Many patients may be cyanotic or anaemic on presentation. Capnography can be useful to quantify genuine hypercapnia in a cyanotic animal. Hypoxia is also commonly seen in patients with respiratory compromise. However it is important to realise the difference between oxygenation of a patient and effective ventilation of that patient.

The ultimate objective of oxygenation is to improve oxygen deliverance to the tissues, specifically the CNS, of your patient. Blood loss or hypovolaemia are other indications for oxygenation of the patient.

Oxygen can be delivered via face mask or flow by oxygen in those patients that will tolerate it (most that bad will). Flush any circuits or anaesthetic machine used first so there is no anaesthetic gas residue left (or breath holding may occur). Otherwise an oxygen tent can be created out of an incubator or propagator or the animals carry cage. A towel, plastic bags or cling film can be used to create a seal. Commercially available units are also marketed for this purpose.

Oxygenation can lead to dramatic recoveries in acute cases. Nebulisation or humidified oxygen can also be provided reducing the risk of desiccation of that patient and providing some fluid therapy. Other agents can also be added to the nebulisation chamber to assist in the stabilisation of that patient, such as antibiotics or mucolytics.

Fluid therapy is important for all critically ill mammals. This is in part due to their high metabolic rate. Offering water in a familiar fashion can help voluntary intake as can offering fresh water in a bowl. Be warned that some patients can collapse or get damp in a bowl of water leading to chilling of that patient. Adding electrolyte and probiotic products to the water can help correct imbalances. Providing fluids via syringe can also be easily performed. Maintenance requirements are quite high around 100mls per kilo per day. This can be provided by parenteral and enteral routes. Urine output and specific gravity can also be a useful guide to the success of fluid therapy.

Obtaining a urine sample is simple in the smaller rodents can be facilitated by placing the animal in a plastic container (when being weighed for example). Handling or restraint or confinement leads to some anxiety and most will produce a urine and faecal sample.

Oral fluids are easily administered and well tolerated by many patients. Commercially available electrolyte and glucose sources can be added to warm tap water. This can be used at home to replace mild losses in small mammals. Care should be taken as there is a risk of reflux or inhalation of the fluids in ill of collapsed patients. Absorption from the GI may also be limited and some patients that are not well handled may well be stressed excessively by this process and alternative methods may well prove more appropriate. The other benefit is you are directly hydrating the GI contents (which can help offset dehydration of the contents) and will also simulate GI activity.

Parenteral fluids should be warmed to body temperature prior to use. An infant bottle warmer with a thermometer ensures that bolus fluids can be given at the optimal temperature or fluids can be maintained in an incubator. Warming drip bags in the microwave or in warm water runs the risk of chilling prior to the fluids being administered.

Subcutaneous fluids are quick to administer, simple to perform, large volumes can be given and are well tolerated by many patients. Depending on the species the range of sites may be limited, for example guinea pigs do not like fluids being given into the scruff. Fluids can be administered more caudally or over the flank. Caution is also required for chinchillas due to the risk of fur slip.

Pain can be present on injection, particularly if large volumes are given. Uptake can be slow if the patient is severely dehydrated. More fractious animals may require anaesthesia for administration.
The addition of hyaluronidase at 1500IU per litre can markedly increase the rate of uptake of fluids and reduce the pain as a result. It can also be used to increase the rate of uptake of subcutaneous drug therapy.

Intraperitoneal fluids are well absorbed, useful if venous access is not possible, can be performed with a conscious patient and large volumes can be given. Caution is to be advised if large volumes are given as this may compress the thoracic volume, lead to organ puncture and impair clotting and inflammatory responses in the abdominal cavity. This is of significance in animals after surgery where healing may be impaired.

These risks can be minimised by drawing back on the plunger to check for organ placement. The caudal quadrant is used with the animal tipped away from the surgeon. The procedure should be performed in a sterile fashion. Restraining the ipsilateral hind leg can assist in correct placement and tense the skin and body wall to ease injection.

Intraosseous techniques are probably underutilised in small mammal practice. This technique is useful for collapsed patients and provides rapid venous support and is easy to perform in all species. Although the lag time is longer than the intravenous route, practically as this technique is simple, rehydration may be achieved more quickly as the time involved in establishing access is much reduced. Ideally the patient should be anaesthetised or local anaesthesia be used over the site. The technique should be performed in a sterile manner to reduce the risk of osteomyelitis. Some patients may not tolerate an indwelling catheter, however these can be utilised in the perioperative period only.

Ideally spinal needles are used as they have a stylet which prevents a bone core forming preventing administration. Alternatively pre loading a syringe and needle with saline and pressurising this can help reduce the risk should spinal needles not be available. Fluids can be given via bolus or via a syringe driver depending on the species being treated. Large rapid bolus administration is painful and should be avoided wherever possible. Care is advised as larger patients can dislodge, bend or remove these needles.

Sites to consider for IO therapy include the proximal femur or proximal tibia. I prefer the proximal tibia as this is a transferrable skill between mammals, reptiles and avian patients.

The proximal tibia should be shaved and prepared aspetically. Lidocaine (quick acting although short length of action) and bupivaciane (slower to act but longer period of anaesthesia) can be given by injection over the site. Care is to be advised so that maximum safe doses are not exceeded.

The joint can be manipulated and the tibial crest identified. The needle should be inserted at the cranial aspect of the joint and down into the medullary cavity. Some force is required initially and the needle inserted using a rotational motion. After initial resistance the needle should slide down the medullary cavity with minimal effort. Typically a needle half to two thirds the length of the bone is used. Once in place the stylet can be removed.

The needle can then be attached to a T port or a syringe driver. Confirmation of correct placement is possible by aspiration (of blood or bone marrow), test injection (if incorrectly placed there will be a fluid swelling at the end of the needle down the leg) or radiographic assessment.

The needle can be bandaged in place and used solely under anaesthesia or after recovery. Ideally the joint should be bandaged in flexion. Removal and replacement should be performed after 2 – 3 days. Another can be placed if needed but for many patients 2 – 3 days is sufficient.

Intravenous routes may be impractical in some of the small species and is typically reserved for ferrets. This route has the advantage of rapid rehydration (even in the severely dehydrated). Careful restraint is needed and access can be difficult due to fractious patients, small fragile veins. Patients may also remove the catheter and chew through any giving set used to administer fluids. Typically a secure catheter is placed with an injection port attached.

Catheters are typically placed in the cephalic vein in ferrets, this is often performed once they are anaesthetised unless they are collapsed.

Crystalloids are often chosen for many small mammals. Hartmanns is ofter the first choice for the majority of conditions. 10 – 15 mls per kilo are given as a bolus. Larger volumes can be given via the intraperitoneal route. Blood transfusions are indicated in small mammals where the packed cell volume has reduced to below 15% in chronic disease. These are not commonly performed generally unless there has been significant blood loss during surgery or with oestrogen toxicity in female ferrets. These are seasonally monoestrus and unless mated the oestrogen leads to bone marrow suppression.

In these cases the jill becomes pale, collapsed and weak. Mucous membranes are pale with poor refill. The PCV can become very low. As there is a marked difference in size between male and female ferrets, males are often used. There is no need for cross matching between ferrets. Donors may need to be anaesthetised to get the volume needed and this should be replaced via the intravenous route. Acid citrate dextrose should be used and a small quantity can be removed from a

blood bag. Donor sites include the jugular or anterior vena cava. Blood can then be administered via the intravenous or intraosseous routes in the female.

Quick guide for emergency fluid therapy in small mammals.

Species	Subcutaneous	Intraperitoneal	Intraosseous	Intravenous
Mouse	2ml	3ml	0.5ml	0.5ml
Hamster	2ml	3ml	0.5ml	N/A
Gerbil	2ml	3ml	0.5ml	N/A
Rat	5ml	10ml	5ml	5ml
Chinchilla	10ml	15ml	5ml	5ml
Guinea Pig	20ml	20ml	5ml	5ml
Ferret	30ml	30ml	10ml	10ml

Supportive nutrition is an important part of the care of a patient with a high metabolic rate. Transit time is fast in ferrets which need to feed quickly. Hysterichomorphic rodents are prone to gastrointestinal slowdown and stasis and supportive nutrition is important to reduce the risk. Higher fibre foods are also important for these species.

Monitoring the weight is important to evaluate the relative need for supportive nutrition. Weight should be monitored once or twice a day. Assist feeding is time consuming and patience is required. Highly palatable foods may reduce the need for assist feeding and are of particular use in fractious patients.

Products used include products such as critical care formula from vetark (for an immediate energy boost) or fibre based products such as oxbow critical are for herbivores, supreme recovery diet or emeraid herbivore diet. For carnivores liquid diets intended for dogs and cats can be used or oxbow produce a carnivore care product as well. Wide bore syringes must be used for fibre based products, however oxbow now produce a fine grind herbivore diet which can pass via a 6 – 8 french tube.

Ferrets typically take food voluntarily and high fat energy dense products can be given in a bowl or by hand. Warming or adding additional fluid may increase intake. Gastric ulceration is also a risk in ferrets and so ranitidine (2 – 4 mg/kg every 8 hours) can be given prophylactically to ferret inpatients. Any products used should be mixed with warmed fluid to increase palatability of the diet.
Fractious rodents can be restrained in a towel whilst being assist fed. Others may take assist feeding voluntarily and this can make life much easier. Many myomorphic rodents can be easily fed palatable products via syringe or spoon. Many owners are capable of providing assist feeding at home. If assist feeding is likely to cause stress, it may be of reduced benefit and providing privacy, analgesia and palatable foods is an alternative. These products can also be offered in a bowl in the cage as an alternative. Smaller rodents can be encouraged to take therapy in other vehicles such as peanut butter or jelly for example.

If longer term nutritional support is required assist feeding can be difficult. In addition animals with painful oral or facial lesions may suffer undue stress with assist feeding.

Gastro intestinal stasis is a potential complication in all hystrichomorphs and administration of prokinetics should be routine for all cases. Gastric ulceration is under diagnosed and ranitidine is therefore the agent of choice for its effects on intestinal motility and its blocking action of H2 receptors reducing the production of gastric acid. Other agents to consider include cisapride, domperidone or metoclopramide.

Analgesia is also important for small mammals. Many of these species are prey species and can hide pain well. Clinical assessment of pain is therefore difficult. Remote observation is ideal to monitor

these species and CCTV or a small window into the ward can allow more subtle behaviour patterns to be seen. Evaluation of the amount of diet eaten, water drunk and faecal and urine output is important. Weighing food and measuring water is important to calculate the actual amounts consumed.

Pre emptive multimodal anaesthesia should be provided whenever pain is assumed to be part of the disease process. Five drug classes are used in small mammals and can be used in combination to provide analgesia. Many of these are part of anaesthetic protocols. Side effects such as respiratory depression or nephrotoxicity have to be considered when selecting the most appropriate agents to utilise.

Non steroidal analgesics should be used as a routine to all patients. These are generally administered once or twice a day and can be given via a variety of routes. The intravenous route is to be preferred when possible. If the hydration status of the patient is of concern (which increases the risk of renal side effects) then this should be corrected prior to administration. These products also have the advantage of palatable oral liquid formulations available for use at home.

Opiod analgesics are often used as part of anaesthetic regimes and are generally used for more major procedures. Agents such as butorphanol, buprenorphine, fentanyl, methadone and morphine are commonly used with pure mu opioids having a greater analgesic effect. Many have a short timeframe of action and require repeat administration. Side effects such as respiratory depression or reduced gastrointestinal motility should be taken into account but are outweighed by the improved analgesia. Buprenorphine is longer lasting (8 – 12 hours) and is absorbed over the mucous membranes via direct dosing or administration in jelly for example. The bioavailabilty of orally administered therapy is low. Many authors recommend a 10 fold increase in the dose. Despite this efficacy is poor.

Tramadol has been subject to some research and currently the serum levels reported are unlikely to provide analgesia. Doses used are 10 mg/kg once a day for 5 mg/kg twice a day orally. Morphine is very useful for short term analgesia for painful procedures or lesions. Fentanyl is combined with fluanisione and marketed in the UK for anaesthesia. If this is to be utilised caution is advised to select appropriate opioids prior to anaesthetic induction to avoid antagonism. The fluanisone also increases sleep time markedly and so other options are often considered.

In recovery morphine can be used to provide further mu opioid analgesia or buprenorphine or butorphanol used to partially antagonise the fentanyl reducing sedative effects but yet providing ongoing analgesia. Dosing regimes vary in the actual dose and dose frequency administered to small mammals and a dosage chart can be useful to tailor species specific doses.

Analgesia of small mammals

Species	Meloxicam	Ketoprofen	Carprofen	Morphine	Buprenorphine	Butorphanol	Tramadol
Ferret	0.1 - 0.2 mg/kg	3 mg/kg	1 – 5 mg/kg	0.5 – 2 mg/kg	0.01 – 0.03 mg/kg	0.1 – 0.5 mg/kg	5 mg/kg PO BID
Chinchilla	0.6mg/kg	1 mg/kg	2 – 4 mg/kg	2 – 5 mg/kg	0.05 – 0.1 mg/kg	0.2 – 1 mg/kg	
Guinea Pig	0.6mg/kg	1 mg/kg	4 mg/kg	2 – 5 mg/kg	0.05 – 0.5 mg/kg	1 - 2 mg/kg	10 mg/kg PO BID
Rat	1 - 2 mg/kg 4 mg/kg PO SID	5 mg/kg	5 mg/kg	2 – 5 mg/kg	0.02 – 0.5 mg/kg	1 - 5 mg/kg	10 mg/kg
Gerbil	1 – 2 mg/kg		5 mg/kg	2 – 5 mg/kg	0.1 – 0.2 mg/kg	1 - 5 mg/kg	
Hamster	1 – 2 mg/kg		5 mg/kg	2 – 5 mg/kg	0.05 -0.5 mg/kg	1 – 5 mg/kg	
Mouse	1 – 2 mg/kg		5 mg/kg	2 – 5 mg/kg	0.05 – 2.5 mg/kg	1 – 5 mg/kg	

Local anaesthetics are under utilised in companion animal practice. Lidocaine and bupivaciane can be used in combination. Lidocaine is quicker acting, but has a short duration. Bupivaciane extends this duration of action. Local anaesthetics totally block pain at the initial site of trauma. These are typically used at 1mg/kg each and mixed in the same syringe (higher doses can be used but it is easy to overdose small patients). For very small patients dilution with saline can increase the volume to be administered allowing a wider field to be anaesthetised.

A typical site these are used in is for blocking the skin and periosteum for intraosseous therapy. Local blocks can also be performed in castrations for example.

Alpha 2 agonists also provide central analgesia and can be used as part of an anaesthetic induction protocol or given at small doses incrementally during a longer procedure. Dexmedetomidine provides some analgesia after the sedative effects have worn off. Medetomidine continues to provide some sedation after the analgesic effects have worn off. Low doses provide some analgesia with reduced cardiovascular effects.

Ketamine also provide analgesia the level of the spinal cord. It is often used in anaesthetic premedication or induction protocols, but also can be given in incremental doses during anaesthesia. It can be used as a continuous rate infusion at very low doses, but intravenous access will be required.

It is also important to avoid sensitisation of prey species which can lead to adrenergic effects and heighten perceived pain. Keeping these species in a quiet ward away from predators, immobilising painful areas, providing hide boxes and gastrointestinal stimulants can all help to reduce pain resulting from injury or disease.

Anaesthesia of small mammals

Prior to anaesthesia providing the owners with some advice is important. This is possible in cases where elective procedures are performed, but may not be possible when an emergency procedure has to be performed. As these species have a high metabolic rate then they should be fed the usual diet until the owner is ready to transport the animal to the clinic. Ideally you should encourage the owners to bring two days worth of the animal's usual diet with them. Feeding this diet can help to limit

the effects of gastrointestinal slowdown whilst at the vets. Animals should be transported at a cool time of day in a secure carrier or box. In addition familiar toys bowls and hides can also help settle in an anxious patient.

Many animals will have specific dietary requirements and providing this is important. Even if the procedure is not elective many owners will come back to the clinic with a platter prepared for their pet whilst it is in the clinic. This allows you to evaluate what the owner perceives as important components of the diet. If the owner does not bring in some of the animal's usual diet then having some food stocks on standby is required.

Assist feeding is an important part prior to anaesthesia. These patients have a high metabolic rate and so starvation is not beneficial, but of course many animals may well be anorexic as part of their condition. If assist feeding is being performed this has to be taken into account when planning anaesthesia.

Starvation is usually not required. Removal of food should occur at the point of premedication in all animals as if heavy sedation occurs they can become sedated with an oropharynx full of food. However, there are a couple of exceptions to this rule. Ferrets for example have a fast gastrointestinal transit time and should be starved for 4 to 6 hours prior to anaesthesia. Typically these have food removed first thing in the morning prior to transport by the owner which is usually sufficient time. Guinea pigs can also be starved for a short time period, primarily to ensure they have a clear oropharynx. Another option is to syringe out their mouth once food has been removed. Having some cotton buds ready to clear the oropharynx in these species is also advised.

A full clinical examination is important as these species hide disease well. Subtle signs of illness may be missed by their owners. This also allows for baseline parameters to be obtained on that individual. Respiratory and dental diseases are common for example.

Elective procedures may require minimal supportive care as the animals are clinically healthy. However sick animals will benefit from a period of supportive care prior to anaesthesia and supportive fluid therapy or nutritional support are important. In mild cases supportive care may be instigated at the point of anaesthesia to minimise stress on the patient.

Intensive critical care is important, however in most cases 1 – 2 days is sufficient. By this point the patient should be sufficiently stable to allow anaesthesia. If this is not the case then the empirical supportive care is not being effective and more specific treatment is required. To achieve this, a diagnosis is important, and anaesthesia will be required in most cases. If the patient continues to be unstable euthanasia may well be indicated. Financial limitations can be a problem when treating small mammals and it is very easy to use up all funds on supportive care without making a diagnosis and treating the animal specifically for its problem. Antibiotics are often abused in these cases as an alternative for a diagnosis.

Patient size is also an important factor to consider. They have a large surface to volume ratio and hypothermia is a risk. Equally if measures used are highly effective then hyperthermia is also a risk during anaesthesia and surgery. Avoiding hypothermia starts with identifying normothermia for that patient on that day and so baseline parameters are required. Reference ranges for example the rabbit rectal temperature can be wide and having a clearly defined pre anaesthetic temperature which should be maintained during and after anaesthesia is important.

Baseline physiological parameters for small mammals.

	HBR	RR	Temp (^{0}C)	Weight (g)
Ferret	200 – 250	33 – 36	38.8	600 – 1200
Chinchilla	100 – 150	45 – 65	37 – 38	400 – 600
Guinea Pig	190 – 300	90 – 150	38.6	750 – 1200
Rat	260 – 450	70 – 150	38	250 – 500
Gerbil	300 – 400	90 -140	37 – 38.5	70 – 120
Syrian Hamster	280 – 412	33 – 127	37.6	90 – 150
Mouse	500 – 600	100 – 250	37.5	20 – 40

It is important to realise that stress and anxiety will influence these parameters. Respiration rate should be taken form observation at a distance and heart rate can often be taken by gently opposing a stethoscope to the side of the patient without physical restraint (Very useful for myomorphic rodents).

Sick patients and those during and after anaesthesia will be unable to regulate their temperature (for example shivering thermogenesis is lost under anaesthesia) and providing a heat source is important. In many cases focal heat is contraindicated as the patient may be unable to move away or towards a heat source. Providing an even heat source over the entire cage is important and monitor rectal temperature to ensure normothermia is maintained. Many small mammals have a poor thermal tolerance and hyperthermia and hypothermia can develop quickly. Most small mammals benefit from an even temperature of 26 – 30 degrees centigrade in the recovery period but quickly need to be transferred to lower temperatures once mobile. Continue to monitor the rectal temperature. Continuous recording devices are helpful as the temperature can be recorded remotely with minimal disturbance of the patient and loss of heat from the incubator for example. Higher humidities can also benefit these patients and reduce the risk of dehydration.

The weight of the patient is important. Weight should be taken twice a day on accurate scales and compared to species ranges and individual records for that patient. It is important to evaluate body condition as well as an animal with marked ascites for example may weigh well but be in poor body condition. This will not only allow for accurate therapeutics but also help in assessing the response of the patient. Owners will very quickly realise the benefit of regular weighing and the first question they will ask when on the phone is ‘what was fluffys’ weight this morning?’ Record the weights on the computer as well for easy access in the consulting room.

Some patients may need to be restrained for weighing but many patients will voluntarily allow themselves to be weighed and owners may even keep accurate daily records at home (particularly if you encourage them). Analgesia is important as the clinical signs of pain may be limited. Bear in mind the need to consider any anaesthetic regime and ensure there is no antagonism between opioids utilised.

Fluid therapy is important and it may be impossible to quantify deficits so many patients are assumed to be dehydrated and a standard rate of fluid therapy provided. This is often 10mls/kg/hour with the route depending on the severity of illness. Hyaluronidase can be added to subcutaneous therapy to speed up absorption. This is expensive so typically a litre bag is made up, dated and placed in the fridge. Fluid is removed in a sterile fashion and the bag discarded at the end of the week.

Pre-emptive antibiotic therapy may be indicated if significant infection is suspected or a contaminated procedure is going to be performed. Finances may preclude culture and sensitivity and if this is the case presumptive therapy for the likely pathogens is required. If a culture is going to be taken give antibiotics afterwards. If the sample is taken at surgery (for example from a dental abscess capsule) then intravenous therapy can be provided to provide immediate cover.

Caution is to be advised regarding the risk of dysbiosis and species specific sensitivities. However ensuring all likely pathogens are treated is important. First line antibiotic cover may include covering for aerobic and anaerobic bacteria and gram positive and negative infections (four quadrant cover). Microbial resistance can be high in some species and if a significant infection is likely our choice of therapy may be altered.

First line choices for ferrets include amoxicillin/clavulanate for example whereas in rodents trimethoprim sulphonamides should be used to avoid dysbiosis. Sulphonamides can be given orally to rodents. Cephalosporins such as cephalexin can be used in ferrets. All of these provide four quadrant cover although resistance is possible. If a significant infection is suspected enrofloxacin (which is licensed) will have good activity against resistant gram negative aerobes, but has no activity against anaerobes. This can easily be achieved by adding metronidazole to extend the spectrum. The advantage is these can be given orally to patients as well as parenterally.

Preparation is the key to any anaesthetic. As small mammals are higher risk patients minimising the length of any anaesthetic procedure is important and identifying all likely items you will need and preparing these in advance is important. Preparing all the emergency drugs is also important. Simple dosing charts can be created and kept in a crash box patient side as an alternative.

Pre medication of patients is often performed and can help with muscle relaxation, analgesia and sedation of the patient (providing all three aspects of the anaesthetic triad). Pre medicants can cause hypothermia however and cause cardiovascular and respiratory depression and so supportive care measures should be implemented at the point of premedication. Waiting until the patient is in theatre prior to dealing with thermal support is a mistake and patients can quickly become markedly hypothermic. Animals are usually placed in a basket and kept in a quiet darkened room and covered with a towel. The time is noted and they are evaluated regularly. They are moved to a heated room to counteract any hypothermia and this avoids subjecting patients who are in the ward being subjected to higher temperatures. Anticholinergics are recommended for use in guinea pigs and chinchillas routinely given the large amount of secretions these animals produce. Ventilation is depressed under anaesthesia. Elevating the thorax and lying the patient in ventral recumbancy can facilitate ventilation of an anaesthetised patient. Operator fatigue is also a problem and the veterinary surgeon dealing with the patient can lead to inadvertent compression of the patient. Instruments, draping and swabs can also have an effect. Sitting down and resting your forearms on the theatre table reduces the likelihood of this occurring and in addition reduces hand tremor and improves dexterity, facilitating fine surgical intervention.

Ferret anaesthesia

Ferrets do not breath hold and mask induction can be performed. They can be lively and premedication is to be advised. If venepucture is required or the patient is critical mask induction can be use and sevoflurane does lead to quicker inductions and recoveries.

Intramuscular administration of drugs is generally not required as subcutaneous therapy works within a few minutes. Dexedetomidine at 0.025mg/kg, ketamine at 5 mg/kg +/- buprenorphine at 0.05 mg/kg all given by subcutaneous injection provides a sedated, analgesed ferret. This should be reserved for healthy individuals. They usually require some gaseous agent as there is often quite marked jaw tone with this combination. However, this level of sedation will provide sufficient immobilisation for non painful procedures such as radiography. Alpha 2 agonists will have cardiovascular effects and so midazolam (0.5mg/kg) can be used as an alternative alongside ketamine 5mg/kg).

Alfaxalone has also been used for ferrets. This has been used with benzodiazepines given by subcutaneous injection. Doses used ranged from 5 – 10 mg/kg. This does require a little gaseous agent to facilitate intubation and a surgical plane.

The administration of isoflurane or sevoflurane by face mask can facilitate intubation. Typically 2 mm endotracheal tubes a required. Local anaesthetic can be applied to the larynx which can be visualised easily. They have a long chest cavity and normal length tubes are suitable. The tube can be secured in the usual fashion. Intubation allows for a secure airway and IPPV if required.

Reversal with atipamezole can be performed if dexmedetomdine has been used at 5 times the dose (half the volume). Waiting for 30 – 45 minutes is recommended.

Myomorphic Rodent anaesthesia

Although intubation has been reported in rodents it is in many cases impractical for the practitioner, however routine intubation of rats using intravenous catheters (with the stylet removed) is possiblke with practice.

Many smaller rodents will require gaseous anaesthesia to be administered via a face mask. This limits the use of IPPV and capnography.

This is of increased concern as these species can have significant underlying respiratory tract pathology. However pulse oximetry or Doppler ultrasound probes can be used to provide a continuous heart rate and oxygenation readout. A critical evaluation of the patient prior to anaesthesia must be undertaken. This must include observation, clinical examination and evaluation of their bodyweight (and comparing this to previous records). Many rodents sold commercially are intensively raised .These may have previous history and have been exposed to a variety of agents. In some cases therapy for underlying respiratory disease may be indicated for 1 – 2 weeks prior to anaesthesia. Typically this is of concern in rats, but other rodents may be hiding underlying diseases as well. Cardiovascular disease is also evident in many species and underdiagnosed.

Myomorphic rodents are small and have a high metabolic rate. So any starvation may be detrimental. These of course may have had limited access to food during transport or may be diseased and anorexic. So food should not be withheld from rodents and in many cases supportive nutrition may be indicated.

Hypothermia is a greater risk in small mammals due to their high metabolic rate and larger surface to volume ratio. Thermoregulatory mechanisms are also lost under anaesthesia. Measuring rectal temperature is very important in small animals.

Induction for most myomorphs is via gaseous induction. Injectable agents can be used as premedicants, but many doses in the literature can reflect a healthy genetically homogenous population (from laboratory medicine), which is totally different to the heterogeneous diseased population seen in clinical practice.

However analgesia should not be overlooked and these can provide some sedation, analgesia and muscle relaxation. They can also have an anaesthetic sparing effect. Given the lively nature of the small rodents and the speed at which agents become systemically available agents, can be given practically at induction.

Chamber induction must be performed correctly. It is important to provide a route for the oxygen carrying the agent to enter and escape from the chamber. Gaseous agents are denser than oxygen or air and as a result sink to the bottom of chambers. Ideally an inlet and outlet should be provided with the outlet at the top of the chamber. Alternatively a large face mask can be used for small rodents. This allows for quick induction and direct observation of the patient. Smaller face masks can be used once the rodent is asleep. Rodent specific models scavenge actively from the face mask reducing environmental contamination.

Hystricomorphic rodent anaesthesia

Hystrichomorphs should be given prokinetic prior to anaesthesia. Degus are generally gas induced given their size but chnchillas and guinea pigs are typically premedicated. Guinea pigs are starved for 1 – 2 hours maximum. This includes the removal of bedding. It is very common for guinea pigs to

have a large material of food within their oropharnyx. This compromises effective ventilation but also hinders examination of the dental arcades for example.

Guinea pigs and chinchillas do breath hold when exposed to a gaseous agent and guinea pigs will lacrimate. As a result premedication of these species is advisable. The use of premedicants can help with all three aspects of the anaesthesia triad and allow for minor procedures such as radiography to be performed. Should gaseous induction be utilised then a chamber or facemask can be used with chamber induction being preferable.

Fentanyl/fluanisone ('Hypnorm') is an option to consider as this is licensed (in guinea pigs and common myomorphic rodents too, but is not licensed in chinchillas). High doses are listed on the datasheet (up to 1 ml/kg) and practically lower doses are used. 0.2 mls per kilo will provide suitable sedation prior to gaseous induction. It does generate a long sleep time due to the fluanisone component (which is not reversible) and the fentanyl is short acting. This means you may be getting mu opioid analgesia when it is not required and the need to top up the fentanyl promptly. Hypnorm can be reversed using a variety of agents. Naloxone can be used in an emergency to reverse the fentanyl, but will of course eliminate any analgesia. Doses used vary between 0.01 – 0.1 mg/kg. More commonly buprenorphine is used to partially reverse the fentanyl reducing the sedative effects but continuing to provide analgesia. Doses used are 0.01 – 0.05 mg/kg. In painful procedures the fentanyl does not require reversal, but morphine can be given 2 hours after its administration to provide ongoing analgesia.

Alternatives include the use of dexmedetomidine 0.025mg/kg with ketamine 5 mg/kg with buprenorphine. Other options include the use of midazolam or acepromazine as alternatives. These will provide suitable levels for radiography or dental examination. These animals should be oxygenated and monitored for hypothermia even if a gaseous agent is not utilised. Isoflurane or sevoflurane can be used to deepen these species. They will still breath hold and struggle to some extent. Both these agents irritate mucous memberanes. Incremental increases in gaseous agent during induction can be used, but do not speed up induction. The use of ocular lubricants and preoxygenation is recommended. Guinea pigs also produce a large amount of secretions and some authors routinely use parasympatholytics (atropine) prior to anaesthesia.

Alternatives in development include supraglottic airway devices (VGELs are being developed to engage over the glottis).

Making anaesthesia safer

IPPV is indicated in all animals that have been intubated. This increases SPO_2 levels (oxygenation) and facilitates removal of carbon dioxide from the lungs $ETCO_2$ (ventilation). Whilst IPPV can be performed by a veterinary nurse, this can be difficult to achieve. In order to achieve effective ventilation the nurse should be focused on this task otherwise there is increased risk of ventilation perfusion mismatching as the pressure and volume and frequency of ventilation is altered markedly. Mechanical ventilators are available and one such unit is an inexpensive pressure cycling ventilator. This can replace a T piece and can be set for IPPV or normal ventilation. This allows IPPV to be instigated and stopped at any point during procedures depending on the requirements of the patient. This unit measures pressure at the end of the ET tube and a value allows a unidirectional flow of gas until the pressure is reached. Pressure and expiratory time can be set. Increasing pressure increases lung volume (based on lung compliance). The aim is to achieve normal chest movements for the animal. It is best to start at lower pressure and observe chest movements of the patient and increase pressure to achieve normal chest movements. Expiration time can be set to provide the normal respiratory rate for the species. Bear in mind that the respiration rate recorded from this patient may be elevated. Recoil of the patient's thoracic cavity facilitates expiration. After a set time period the value closes and inspiration occurs. As the pressure is recorded at the end of the ET tube caution is required for patients with small ET tubes. In these cases if a high flow rate is used pressure can build up and the value is opened, before the patient is sufficiently inflated. This can be corrected by lowering the pressure, then the flow rate and slowing increasing the pressure again. It is important not to alter flow rate whilst the patient has a high pressure setting as hyperventilation is possible. This problem usually occurs with patients with endotracheal tubes of less than 2mm in diameter. Ideally the flow rate should be geared to keep inspiration within 1 – 2 seconds and expiration time depends on the normal respiratory rate you would expect for the species. IPPV when used appropriate

facilitates and stabilises anaesthesia, ensures inhalational agent delivery to the lungs, oxygenation of the lungs and removal of carbon dioxide from the alveoli. Caution is needed to reduce the negative effects of mechanical ventilation on blood flow and the risk of lung trauma.

Monitoring patients under anaesthesia is important and is typically performed by the veterinary nurse. However, particularly if IPPV is used, the outward parameters to monitor anaesthesia can be limited in small mammal patients and using monitoring devices can help to reduce both morbidity and mortality under anaesthesia.

Reflexes typically monitored to evaluate the plane of anaesthesia include toe pinch/withdrawal reflex, heart and respiratory rate and rhythm, eye position, mucous membrane colour and refill time, voluntary movement and response to surgical anticipation. Ideally the last two should be pre empted and the plane altered accordingly to the level of surgical stimulation.

Pulse oximetry has been used in many species. This measures the oxygenation of the patient by the oxygen saturation of the haemoglobin molecules (SpO_2). This correlates well with the arterial oxygen (PaO_2) levels. The machines are based on the human oxygen haemoglobin dissociation curve. Normal values should be 96% or more. Probes can be difficult to attach to some of the smaller patients. Caution is to be advised if alpha two agonists are utilised as poor peripheral perfusion can lead to improper functioning of the unit. Pulse oximetry does not provide information on blood flow of the oxygenation of the animal's tissues (which is in fact the important objective). Many machines are manufactured for human use and are unable to detect heart rates over 250 BPM. There are machines on the market, which can record rates up to 350 BPM. This can still be exceeded by small mammals, but is unusual in the clinical setting. Pulse oximetry does not tell us anything about the effective ventilation of the patient. They are also quite insensitive. An example helps to identify the short comings of pulse oximetry. If you had a pulse oximeter attached to your finger and then held your breath it is likely you would become hypercapnic and have to breathe again prior to the pulse oximeter detecting a fall in SPO2 levels. A capnograph for example would have immediately detected the breath holding and the subsequent hypercapnia as a result. For this reason capnography is my preferred option for anaesthetic monitoring in small mammals when ever possible.

Capnography measures effective ventilation of the patient and ventilatory failure is the most common cause of morbidity and mortality under anaesthesia. Capnography is highly sensitive and reacts far quicker than pulse oximetry in an emergency situation. Capnography measure the end tidal CO_2 levels. It depends on the rapid exchange of gases between the lungs and the circulation and $ETCO_2$ correlates well with $PaCO_2$. As this is a real time monitor and is highly sensitive. The only delay is the time taken for the expired air to reach the monitor. A capnograph provides consistent reliable real time values. They can be used on anaesthetised and conscious patients (to help in the clinical assessment of hypercapnia for example as mucous membrane colour is unreliable) and all species have similar $ETCO_2$ values. The display is simple to use and interpret. Side stream units are most commonly used (as opposed to mainstream units which are more costly) it is important to ensure these have a low flow rate to avoid interrupting the gas flow for small patients. Small adapters can be utilised to minimise dead space. Respiratory rates up to 120 breaths per minute can be recorded. A capnograph and a capnogram are different. A capnogram provides a record of $ETCO_2$ which is calculated by the difference between peak and trough values. A capnograph displays a real time waveform of the level of CO_2 in the gas flow allowing the clinician to evaluate underlying pathology, the respiratory rate and the $ETCO_2$. Capnograms calculate the ETCO2 based on the difference between peak and trough values so a patient that is hyperventilating and re breathing will have incorrect results displayed. Capnography can be used in patients on a facemask. Typically the probe is placed at the end of the nares within the facemask to get some idea of the CO2 wavefrom. At this distance it is unreliable but at least demonstates CO2 output with each breath.

Doppler flow monitors are also very useful. These have become increasingly available in clinical practice for blood pressure monitoring. They are easy to apply and can be used to obtain an audible continuous readout of the heart rate and blood flow in small mammals. The rhythm and rate can be assessed. These can be taped in place over the heart or a peripheral vessel. Flat probes are best for these purposes although a pen style of probe can be used for intermittent evaluation. The heart rate can be calculated or the anaesthetist can concentrate on the rhythm and pick up on subtle changes in rate in real time. The second advantage of these units is for indirect blood pressure monitoring in patients of sufficient size. All anaesthetic agents will create some hypotension during anaesthesia

(due to reduced baroreceptor reflex and vasodilation). Measuring blood pressure will allow fluid therapy (particularly if colloids are utilised) to be titrated. Mean arterial pressures recorded can range between 70 – 170 mmHg. The fore leg is typically used. The cuff is placed high up on the limb with the doppler probe below. Blood flow is detected and the cuff inflated until sounds are lost. The cuff is then slowly deflated until the flow is audible again. This is repeated three times and the average taken as the mean arterial pressure (MAP). Cuff size is important to obtain consistent results.

ECG recording can also be performed. This is more time consuming and difficult to get consistency, particularly if electrical items are being used (such as radiosurgery) can lead to electrical interference and so this is less commonly used as a monitoring tool for small mammals. However specific patients, based on their presentation, may well require ECG recording as part of a safe anaesthetic regime.

The recovery period is a critical part of the regime. Once gaseous agents are turned off continued respiratory support is indicated as respiratory compromise is common during recovery. Reversal agents can be administered to speed recovery, but this has to be balanced with the analgesic effects of agent such as the alpha two agonists, opioids or ketamine for example, which may be beneficial in the immediate post operative period. If low doses have been used reversal may not hasten return to mobility and feeding.

Atipamezole is regularly used for reversal of alpha two agonists. In ferrets atipamezole is given at the dose rare of 2.5 times the dose of medetomidine (half the volume) in all other cases 5 times the dose is administered (the same volume). Subcutaneous therapy is often used. In ferrets it is best to wait for 45 minutes prior to reversal. Naloxone should be available to completely reverse any opioids. This will reverse any analgesia and is usually reserved for an inadvertent overdose or where complications have occurred. Buprenorphine is a partial mu agonist and can be used to reverse some of the effects of pure mu opioids. Buprenorphine also has greater affinity for receptors and so displaces other opioids more readily. Its advantage is greater analgesia of a longer duration.

Jaw tone and movements is an indication recovery is imminent and the endotracheal tube (if present) can be removed. In many cases the tube stimulates the patient and once removed many patients will tolerate further oxygen by mask. The return of shivering thermogenesis is also an indication of recovery. It is important to plan for recoveries and have a warmed recovery cage prepared. This should be an incubator warmed to 28 degrees centigrade. It is important to continue to monitor respiration, heart rate and rectal temperature (if tolerated) throughout this period. All measures used to maintain normothermia during the anaesthetic should be maintained during the initial phase of recovery.

Once normothermic the incubator is turned off with the animal left inside. If their temperature is held they are moved into their usual cage. Further monitoring is needed to ensure the temperature does not drop further once thermal support is removed. Food and water should be available once the animal is coordinated (be aware that some will get wet if bowls are provided which can chill the patient). It is important to provide hide boxes, towels, blankets and nesting material where appropriate to allow these animals to hide. The ward temperature could be elevated slightly in the initial period or maintained overnight. Digital fan heaters are available with a thermostat which can be used to keep a ward at an even overnight temperature. Feeding, drinking, faecal and urine output should be monitored post anaesthesia. Fluid therapy and analgesia will need to be continued during this period. Nutritional support is important for those species with a high metabolic rate. Gastrointestinal stimulating drugs should also be continued. Improving motility will reduce pain (from gas formation and ileus) and analgesics will also reduce pain (the benefits of this outweighs any effect on bowel motility). Antibiotic usage also must be tailored to the species and need based on clinical presentation. Ranititidine is used as standard as a prokinetic in hystricomorphic rodents (4 mg/kg TID). In ferrets it is used for its protective function against ulceration. This is continued until the animal is eating well and passing normal faecal pellets. Assist feeding with a suitable recovery diet also stimulates motility.

Emergency drug therapy

Agent	Dose
Atropine	0.04 – 0.1 mg/kg (up to 3 mg/kg rabbit)
Glycopyrolate	0.01 – 0.02 mg/kg
Doxapram	2 – 10 mg/kg
Adrenaline	1:10,000 give 0.5 ml
Dexamethasone	0.5 – 2 mg/kg