EXOTIC SMALL MAMMAL ANESTHETIC TECHNIQUES

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Exotic small mammals (also known as pocket pets) are challenging creatures to work with anesthetically. When working with dogs and cats, it is usually easy to intubate, place an intravenous catheter and provide fluid therapy, hook up an electrocardiograph (ECG), place a blood pressure cuff, and keep track of the core body temperature. This can be much more difficult with many exotic small mammals. How do we compensate for the lack of monitoring? We may not be able to, but we as anesthetists can do our best to monitor the patient under anesthesia and be able to anticipate the needs of the patient.

PREPARING FOR THE ANESTHETIC PROCEDURE

Whether the animal needs to have general anesthesia because of a routine procedure, such as a rabbit spay, or it has come into the clinic on emergency for a fractured limb, the same protocols and procedures are usually followed. A general rule of thumb (although there are exceptions) is that all animals under anesthesia obtain some sort of fluid therapy whether it is intravenous (IV), intraosseous (IO), or subcutaneous (SC); IV catheters are preferred over other fluid therapy routes. Emergency drugs should be pre-calculated and drawn up prior to an animal going under anesthesia for any length of time. This may seem wasteful because most of the time the drugs are not used and need to be discarded once the animal has fully recovered. Drugs are always pre-drawn prior to starting the anesthetic procedure because under many circumstances, there is not a lot of time to figure out doses and draw up drugs once the patient starts doing poorly under anesthesia. A cheat chart can be hung on the wall for quick reference or a spreadsheet program can be created. Spreadsheets are helpful as one can be created for each major group of animals commonly seen in your practice (e.g., chinchillas, rabbits, rats). The spreadsheet should remain simple and contain the common drugs used during emergency situations. Spreadsheet programs are nice because they can be used over and over again. By just inputting the patient’s weight, the program will calculate the proper drug doses.

It is important to have everything set up and within hand’s reach before starting the anesthetic procedure. This includes all equipment that may be needed such as an ECG, blood pressure cuff and sphygmomanometer, ETCO₂, pulse oximeter, endotracheal tube, catheter supplies, fluids, drugs, syringe pump, etc. Being prepared will help the procedure
move along in an organized fashion. It is important to have pediatric items such as 26- and 24-gauge IV catheters, 3.5-mm uncuffed endotracheal (ET) tubes and smaller (18- to 14-gauge catheters can be adapted into tiny ET tubes), mini volume IV lines, etc.

Fluid therapy routes in small mammals under anesthesia include subcutaneous, intravenous, and intraosseous administration; IV and IO fluid therapy can be difficult in some small rodents. Vessels used for IV catheterization include the cephalic and lateral saphenous in most small mammals. In rabbits the auricular (lateral ear) veins can be used, but the cephalic and lateral saphenous should be attempted first. The lateral auricular veins are small, can blow easily, and in rare cases, the ear can slough when certain drugs are given peri-vascularly. In rats, the lateral tail veins can be used to place an IV catheter. If IV access is not possible due to poor perfusion, an IO catheter can be placed. It is best to use a spinal needle when possible because there is a lesser chance of the lumen getting clogged with a bone core when being placed. A regular hypodermic needle can be used when needed. The IO catheter is usually placed in the proximal femur or the tibial crest if the patient is large enough. Common fluids that can be given include crystalloids, such as Lactated Ringer’s Solution (+/- dextrose) and sodium chloride, and colloids, such as Hetastarch. With the exception of Hetastarch (dose will vary by species), anesthetic maintenance in small exotic mammals for most fluids is 10 mL/kg/hour. This is the same rate used with dogs and cats. It is important to monitor the patient when on IV or IO fluids as some patients can become fluid overloaded. If the anesthetic procedure is long, the fluids can be given at 10 mL/kg/hour for the first hour and then dropped down to 5 mL/kg/hour to help prevent excess fluid overload. Fluid rates should be adjusted based on the needs of the individual patient. This is especially true if giving fluids via the IO route. If SC fluid therapy is chosen then fluids are generally given at a rate of 60 mL/kg/day. Subcutaneous fluids are only given when a procedure is very short and an IV catheter is not necessarily needed (e.g., ultrasound or radiographs under anesthesia) or an IV catheter is unable to be placed (e.g., the animal is very small or the vessels have been blown).

**ANESTHETIC MONITORING EQUIPMENT**

Monitoring exotic small mammals under anesthesia is extremely important. Many of the patients being anesthetized weigh between 30 grams and 6.5 kg (most weighing less than 3.5 kg). Due to the small size of these patients, what type of monitoring equipment can be used? Most of the same equipment used on small cats can be used on rabbits and ferrets as well. For major surgical procedures or when anesthetizing a high-risk patient, monitoring equipment such as an ECG, Doppler and sphygmomanometer with blood pressure cuff, temperature probe, pulse oximeter, and capnograph should be used when at all possible. When possible, an arterial catheter should be placed to monitor direct blood pressure and monitor blood gases. If anesthetizing a patient for a minor surgical procedure or if the patient is considered low risk, the minimal monitoring equipment should include, but is not limited to temperature probe or thermometer (if patient is not too small), Doppler with sphygmomanometer and blood pressure cuff, +/- pulse oximeter
and capnograph. If the patient is too small to obtain a true blood pressure reading, the anesthetist should look for trends. For example, if the blood pressure cuff is too large (commonly happens with small rodents) and the blood pressure consistently reads a systolic of 70, but then suddenly drops to 40 you know that either the cuff slipped or came off or you are seeing a downward trend indicating hypotension. Looking at trends can be a helpful and informative way to manage your patient.

As a side note, it is important to use pediatric equipment for most exotic small mammals being anesthetized. Using pediatric equipment will give more accurate results. I have found that the Digicare Life Window is one of the best monitors on the market today for exotics. It reads ECG, BP (direct and indirect), Temp, RR, ETCO$_2$, SPO$_2$, etc. This machine also works well on dogs and cats so it is multifunctional.

**ANESTHETIC INDUCTION**

Most animals are given pre-medications prior to being placed under general anesthesia, although this will depend on the preference of the clinician and will vary with each patient. Common premedication drugs used in exotic small mammals such as rabbits and rodents include drugs such as glycopyrrolate, butorphanol, midazolam, ketamine, buprenorphine, and full mu opioids such as oxymorphone. A current exotic animal formulary should be consulted before administering any drugs. Exotic small mammals are not just little dogs and cats. Most drugs are given directly into the muscle because uptake is generally quicker, but most drugs can be given subcutaneous as well. After pre-medications have been given, the animal should be placed in a clean, empty cage while the drugs become effective. This generally takes 10 to 20 minutes, but depends on the drug combination used and the disposition and mentation of the patient. After the pre-medications have taken effect, general anesthesia can be induced.

There are a few different techniques that can be used to induce anesthesia. When at all possible, it is suggested to pre-place a catheter, pre-oxygenate the patient and induce anesthesia with injectable drug combinations such as ketamine and diazepam or ketamine and midazolam (same technique as a dog or cat). Propofol can be used as an induction agent in ferrets, but is not suggested in other small mammals as they are often difficult to intubate and apnea is a potential side effect of injecting propofol too quickly. If a catheter cannot be pre-placed (sometimes the case in small exotic mammals such as guinea pigs, chinchillas, and other small rodents), the patient can either be masked down or placed in an induction chamber using a gaseous inhalant anesthetic such as isoflurane or sevoflurane. A small mask can be placed over the face similar to a dog or cat, or the animal can simply be placed in an induction chamber. The heart and respiratory rates should be monitored from the time the animal is induced until the time the animal awakes from anesthesia. This will help prevent any anesthetic-related problems including death. In some cases, placing a face mask on the patient’s head can be stressful; therefore, an induction chamber should be used. Induction chambers are great because they potentially provide a less stressful induction to the patient. Unfortunately, the heart rate cannot be
monitored during the time induction is taking place. There are pros and cons to either induction method. The induction method should be chosen based on the needs of the patient. Assessing the plane of anesthesia is accomplished in the same manner as a dog or cat. The most common forms include eye position, jaw tone, palpebral reflex, and toe pinch.

NOTE: It is important to remember that most exotic small mammals are prey species. Holding them down for a mask induction can be extremely stressful and can increase their chances of death. Good sedation prior to induction is imperative!

Once the patient is induced, placing the catheter (if not already placed), inserting an endotracheal tube (if possible), and hooking up the monitoring equipment should be accomplished. This can become a tricky situation because attaching all of the monitoring equipment, placing a catheter, and inserting the endotracheal tube should not take longer than the procedure the animal is being anesthetized for. It is then up to the clinician and anesthetist to decide what the most important tasks are, how they should be accomplished, and then set a time limit. It is easy to spend 45 minutes hooking up five different pieces of equipment and then only spend 20 on the actual procedure itself. This is something that should be avoided. This is also a good time to make sure the animal is on a heating source such as a heating pad or hot air blanket. Small mammals have a very large surface area to volume ratio which means they will lose body heat very fast. Keeping them warm is an essential part of a speedy recovery.

Intubation can be extremely difficult if not impossible in some species of exotic small mammals. Intubating ferrets are among the easiest because they are very similar to cats. Generally a 2.0- to 3.5-mm sized endotracheal tube is used. The use of lidocaine will help with laryngeal spasms. Rabbits can be difficult to intubate, but luckily there are a few techniques that can be used to accomplish this task. Due to the oral anatomy, the rabbit’s mouth does not open very wide. The cheeks are fleshy, which also make it hard to see the tracheal opening. Blind intubation is usually the easiest way to intubate a rabbit. This can be done using an esophageal stethoscope or simply putting your ear to the endotracheal tube and slowly moving the tube towards the tracheal opening. Once the tube is close to the trachea, you will be able to hear and feel the respirations on your ear. With practice, you will be able to tell when the tube is right on top of the tracheal opening. Lidocaine should always be used to help with laryngeal spasms. I like to place the endotracheal tube just above the glottis. Once I am near the tracheal opening, I insert a Tomcat catheter hooked to a 1-cc syringe down the endotracheal tube. I administer a small amount of lidocaine (2 mg/kg) onto the tracheal opening. This will take about 1 minute to become effective and the tube can then be more easily placed into the trachea. This technique greatly increases your chances of successfully intubating the rabbit. When attempting to intubate a rabbit, make sure the head is hyper-extended. This will help the tube slip into the trachea.

Rats can be intubated using a very small laryngoscope and usually an 18- or 16-gauge IV catheter modified into an endotracheal tube. Mice, hamsters, guinea pigs, and chinchillas
are very difficult to intubate. In general, these species are not routinely intubated, but an endotracheal tube should always be ready in case the animal goes into respiratory arrest. A rigid endoscope can be used to intubate many exotic small mammals, including rabbits, guinea pigs, and chinchillas. This technique is usually reserved for trying to intubate guinea pigs and chinchillas, but it can be used with rabbits that may be difficult to intubate. Any intubated animal can be placed on a positive pressure ventilator or hand ventilated as needed. This will ensure the animal is breathing normally.

ANESTHETIC MAINTENANCE

Most commonly, exotic small mammal patients are maintained on inhaled anesthetics such as isoflurane or sevoflurane. The percentage will vary based on the patient, procedure being performed, and the pre-anesthetics used. When appropriate (airway procedures, increased intracranial pressure, etc.), a propofol constant rate infusion (CRI) can be used to maintain anesthesia in intubated patients.

SUPPLEMENTAL DRUGS

Rabbits are notorious for having extremely low blood pressure when being maintained on inhalant anesthetics. It is important to try and reduce the need for increased percentages of gas anesthesia. When possible, the anesthetist should consider using pain management techniques, such as administering a fentanyl and/or ketamine CRI, placing an epidural, and/or employing other local blocks when possible. These techniques are generally the same techniques used with dogs and cats. Exotic small mammals recover much faster when provided with appropriate pain control. If hypotension continues to be a problem and the inhalant anesthesia cannot be decreased, a fluid bolus and/or a dopamine CRI should be considered.

RECORD KEEPING

It is important to keep some sort of anesthetic record for each patient that is anesthetized. The anesthetic record should consist of a piece of paper that can be placed into the patient’s normal record and be able to be reviewed if the patient is anesthetized again in the future. Anesthetic records should be simple and easy to read. At a minimum, the record should have a space for weight, heart and respiratory rates prior to and during anesthesia; pre-medications used; what the animal was induced with and maintained with; how much fluids were given if any; the temperature of the animal; what the blood pressure was; the size of the endotracheal tube used, if any; and a comments section. Vital signs should ideally be recorded every 5 minutes.

BLOOD TRANSFUSIONS DURING ANESTHESIA

Blood transfusions can be given when necessary. A general rule of thumb is to consider transfusing when the animal’s packed cell volume (PCV) is between 15% and 20%. The
limiting factors include vascular access and obtaining a safe blood product. Whole blood can be given for transfusion. A donor of the same species is needed to obtain fresh whole blood. The donor animal should be deemed healthy by performing a complete physical examination and running a complete blood count (CBC) and biochemistry panel. If there is only a limited amount of blood, the CBC should be chosen over the biochemistry. Blood drawn from the donor should be collected in a syringe containing the anticoagulant citrate-phosphate-dextrose-adenine (CPDA). The dilution will depend on the species being worked with, but in general is a 1:9 dilution with 9 parts blood and 1 part CPDA. Cross-matching the donor to the recipient should be performed when possible. This will help reduce the risk of the recipient cross-reacting with the donor’s blood once it has been given. It is important to always use a blood filter when performing a blood transfusion on any patient. When giving a blood transfusion during an anesthetic procedure, it is important to first flush the IV line with sodium chloride (NaCl 0.9%). Once the line is fully flushed, the transfusion can be started.

SPECIALIZED PAIN MANAGEMENT TECHNIQUES

Controlling pain in exotic small mammals is extremely important. Epidural anesthesia and CRIs of analgesic drugs are commonly used in dogs and cats. Many people do not use these techniques in exotic small mammals but they should be taken into consideration as a form of pain management. The species, type of procedure, and status of the patient must be taken into consideration prior to administration. Epidural anesthesia and analgesia should be considered for painful abdominal surgery, orthopedic procedures (especially of the pelvic limbs), and thoracic surgery. Epidural placement is performed in the same manner as in dogs and cats. The patient is placed in sternal recumbency. The spine should be straight and symmetric. The hind legs should be pulled forward and positioned against the sides of the abdomen. Pulling the legs forward will open the epidural space. The wings of the ilium should first be palpated. The lumbosacral space can be palpated between vertebral bodies L7 and S1. Prior to placing the epidural needle, the area should be shaved and aseptically prepared as you would for a surgical procedure. Sterile gloves must be worn when administering the epidural. A 25- or 22-gauge spinal needle should be used because it contains a stylet. The spinal needle should be placed on midline, perpendicular to the skin and slowly inserted into the epidural space. A pop will often be felt as the needle passes through the ligamentum flavum and enters the epidural space. If you hit bone, you have gone too far and need to back the needle out a little bit. A sterile glass syringe containing a small amount of air should be placed on the spinal needle and injected into the space. If the air injects easily then you are in the correct spot. If there is a vacuum on the syringe you are not in the correct space and you need to reposition. You can also use a technique called the hanging drop technique. Once you have placed the spinal needle into the epidural space, you can place a drop of saline onto the opening of the spinal needle. If the drop is sucked into the hub of the spinal needle, you are in the correct space, if the drop of saline is not sucked into the spinal needle, you are not in the correct spot. Once you are in the correct space you can place the syringe with the drugs in it onto the spinal needle. Always aspirate back to ensure there is not any
blood or spinal fluid. If blood is aspirated you need to pull out and start over. If spinal fluid is aspirated, you should only deliver about 1/4 of the initial calculated dose. Common drugs used for epidural administration include preservative free morphine, lidocaine, bupivacaine, and buprenorphine.

**NOTE:** Epidural anesthesia/analgesia should not be administered if the patient is septic or has signs of pyoderma or a skin infection around the epidural site.

### Common Drug Dosages for Epidural Administration

- **Preservative Free Morphine** – 0.1 mg/kg diluted to 0.33 mL/kg with sterile saline, administered epidurally (maximum volume of 6.0 mL)
- **Preservative Free Buprenorphine** – 12.5 μg/kg diluted to 0.33 mL/kg with sterile saline administered epidurally (maximum volume of 6.0 mL)
- **Preservative Free Lidocaine** – 0.5–1.0 mg/kg diluted to 0.33 mL/kg with sterile saline administered epidurally (maximum volume of 6.0 mL)
- **Preservative Free Bupivicaine** – 0.5–1.0 mg/kg diluted to 0.33 mL/kg with sterile saline administered epidurally (maximum volume of 6.0 mL)
- **Preservative Free Morphine and Bupivicaine** – 0.1 mg/kg of preservative free morphine mixed with 0.5–1.0 mg/kg bupivicaine (maximum volume 6 mL)
- **Preservative Free Buprenorphine and Bupivicaine** – 12.5 μg/kg buprenorphine mixed with 0.5–1.0 mg/kg bupivicaine (maximum volume 6.0 mL)

**NOTE:** It is important to note that the maximum dose of lidocaine and bupivicaine should not exceed 2 mg/kg and 1 mg/kg, respectively. You must take into account any lidocaine and/or bupivicaine administered not only in the epidural, but also given in other local blocks, such as ring blocks, line blocks, or even small amounts administered onto the tracheal opening to prevent laryngospasms.

Delivering a constant rate infusion(s) during general anesthesia is an excellent way to provide additional analgesia to the patient. Common drugs used for analgesic CRI's include ketamine and fentanyl. Using one or both of these drugs not only helps provide additional analgesia, but they may also help reduce the percentage of gas anesthesia needed to keep the patient in a surgical plane of anesthesia [minimum alveolar concentration (MAC) reduction]. Reducing the amount of gas anesthesia has many benefits including helping with hypotension commonly seen in rabbits while anesthetized with inhalants such as isoflurane and sevoflurane. If fentanyl is used as a CRI, the patient should be intubated and placed on intermittent positive pressure ventilation (IPPV) as this drug can cause severe respiratory depression.

**NOTE:** Patients should be intubated and provided with IPPV via mechanical or manual ventilation if placed on a fentanyl CRI due to potential respiratory depression.
In many instances, analgesic CRIs require a loading dose given at the onset of CRI delivery. A loading dose will quickly increase the drug plasma concentration levels, enabling the low dose CRI to become effective quickly.

**Common Drug Dosages for CRIs**

- **Ketamine CRI** – Loading Dose: 0.5–1 mg/kg
  
  CRI - 10–20 μg/kg/min

- **Fentanyl CRI** – Loading Dose: 5 μg/kg
  
  CRI: 0.5–1.0 μg/kg/min (anesthesia)
  
  CRI: 0.05–0.3 μg/kg/min (post-op)

If you are inducing general anesthesia with ketamine or fentanyl, you can use your induction dose as your loading dose as long as you start your CRI within a few minutes of induction.

**CALCULATING A CONSTANT RATE INFUSION**

Calculating a CRI is very easy once you understand what formula to use. For example, let’s say you are about to anesthetize a rabbit that requires a fracture repair of the right femur. How would you calculate a CRI of fentanyl?

The formula for calculating a CRI is as follows:

\[
\frac{([\text{Patient’s weight}] \times (\text{Dosage of the drug}) \times (*\text{Time factor}))}{\text{Concentration of the drug}}
\]

*The time factor for this equation is 60 minutes/hour

Let’s say the rabbit weighs 2.0 kg and the dose of fentanyl that we are going to administer is 0.7 μg/kg/min. Since my dose is given as μg/kg/min, I will want to convert this to mL/hr. The concentration of fentanyl is 50 μg/mL. I now have all the information I need to calculate the CRI. How will I calculate this?

\[
\frac{((2.0 \text{ kg}) \times (0.7 \mu\text{g/kg/min}) \times (60 \text{ min/hr}))}{50 \mu\text{g/mL}} = 1.68 \text{ mL/hr}
\]
This standard equation can be used for other CRI’s such as ketamine and dopamine.

RECOVERY AND POSTOPERATIVE PAIN MEDICATIONS

The patient should continue to be monitored until it is awake and extubated (if intubated). If a painful procedure was performed on the animal, postoperative pain medications should be given to the patient. Common postoperative pain medications include opioids, such butorphanol, buprenorphine, and full mu opioids, and NSAIDs, such as meloxicam. Remember that butorphanol and buprenorphine are not drugs that should be used for severely painful procedures as they do not provide appropriate pain relief (This is true for dogs and cats, too!). If the patient was on a fentanyl CRI during anesthesia the CRI can be maintained at a lower dose post-op which provides excellent pain management post-op. This is true for a ketamine CRI, too. Regardless of the pain medication chosen, it should be given as needed postoperatively to provide appropriate pain relief to the patient. Exotic small mammals recover poorly and often will become anorexic when in pain. This can be a death sentence for the herbivorous mammals, such as rabbits, guinea pigs, and chinchillas. The IV catheter should not be removed until the animal is fully awake and is no longer needed. Always remember that pain management plays a key role in a speedy recovery for the exotic small mammal patient!