Small Mammal Anesthesia and Critical Care:
Tips and Tricks of the Trade
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Preparation
A clinic or veterinary facility planning to see critical exotic animal cases needs to do some homework. There are hundreds of different exotic pets on the market today. It is vital to understand that different species of rodents, birds, or reptiles are not different “breeds” as in cats or dogs, but are completely different species, often with very different requirements. Any clinic that plans to see exotic patients should have an understanding and familiarity with the species they plan to see—and a series of reference sources for the “surprise” species that happen through the door.

Equipment
In a well out-fitted small animal practice, there usually exist many of the tools needed for exotic mammal emergencies. This is a very basic list of equipment that should be available for exotic mammal cases:
In the clinic:

- Appropriate housing
- Safe and escape proof exam rooms
- Laboratory support equipment
- Gas anesthesia machine with both rebreathing and non-rebreathing circuits

Supplies

- Endotracheal tubes—size 1-6mm, cuffed and uncuffed
- Gas inhalant—isoﬂurane or sevoflurane (or both!)
- Oxygen mask/anesthetic induction mask in a variety of shapes and sizes
- Anesthetic induction chamber, small and large
- Appropriate heat sources—water blanket, warm air convection unit, etc.
- Small, rapid, digital thermometer
- Monitoring equipment—at a minimum a Doppler flow detector with gel, blood pressure cuffs, and a sphygmomanometer, and a pulse oximeter with rectal and lingual probes
- Syringe pump or fluid pump capable of accurately delivering small volumes
- Electronic digital scale
- Incubators in several sizes
- Oral speculums in a variety of sizes
- Restraint/exam items—towels, bags, sturdy gloves
- Client education/appropriate husbandry materials
- Species appropriate food for patients being treated

Examination and triage
Critically ill exotic mammals may not be able to cope with handling or intensive examination. If the animal is not in imminent danger, perform a quick visual-only exam and place the animal in a dark, quiet, warm area for 5 or 10 minutes. This allows the patient to “decompress” before examination. As much of the physical examination as possible should be done with the animal in the carrier.

Once the patient is removed from the carrier, they should be placed on a clean, dry, skid-proof table. All equipment needed for the examination should be immediately on hand. Thoracic auscultation, examination of the ears, eyes, nose, mouth, abdominal palpation, hydration assessment, and weighing of the patient should all be performed quickly. If the patient is distressed, the exam may need to be performed with pauses to allow the patient to recover. Use these breaks to obtain as much information as possible from the owner on the patient’s diet, husbandry, exposure to toxins, or description of traumatic episode. Providing flow by oxygen is never a wrong choice in any critical patient. Patients in respiratory distress may need to examined very quickly and place in an oxygen cage.

The ABCs
Airway
Examine the airway with direct visualization if the patient is tractable enough. If the animal is awake and alert enough to truly resist an oral exam, airway assistance may not be necessary at that moment, though vigilance is still required as the situation can deteriorate. A patient that is severely obtunded or unresponsive may not be to protect the airway. If there is any question, intubate the animal, or if intubation is not feasible, perform a tracheostomy or tracheotomy. Species specific intubation techniques will be described later.
**Tracheostomy**

To perform the tracheostomy, the patient should be placed in dorsal recumbency and the area aseptically prepared. If possible, local anesthetic should be injected over the site and a small incision made on the ventral midline, in line with the trachea and just above the larynx. Dissection through the paired strap muscles and an incision through the tracheal rings is performed. An appropriately sized endotracheal tube or catheter is placed and secured.

**Facemask**

In the event that a patient cannot be intubated and a tracheostomy is not able to be performed, breaths can be delivered using a tight-fitting induction mask placed over the mouth and nose. Ventilation is performed with high pressure breaths using the reservoir bag. This method is not a guaranteed way to adequately ventilate a patient and should be looked at as a last resort.

**Breathing**

Once the airway is secure, either with intubation or self-ventilation, oxygen should be supplied. If the patient is not intubated, oxygen therapy can be provided through a face mask, flow by administration, an oxygen chamber, or nasal cannula. If the patient is intubated, evaluate whether they are breathing on their own or required assisted ventilation with an anesthetic circuit or an ambu bag. Typical ventilation rate is 10-20 breaths per minute, not exceeding 10cmH2O.

**Circulation**

Auscult the patient and palpate peripheral pulses. Assess rate, rhythm and pulse quality. If possible place a Doppler flow detector and cuff or use a reliable non-invasive blood pressure monitor to determine blood pressure. The goal is to have a systolic pressure above 90mmHg. Evaluate mucus membrane (normal=pink) and capillary refill time (normal=1-2 seconds). If any of the circulatory parameters are below normal, fluid therapy may be indicated. Intravenous and intraosseous catheters can be placed in the following species/locations:

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<tr>
<th>Ferrets</th>
<th>Intravenous Catheter</th>
<th>Intraosseous Catheter</th>
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<tr>
<td></td>
<td>Cephalic</td>
<td>Tibial crest</td>
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<tr>
<td></td>
<td>Lateral saphenous</td>
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<td></td>
<td>Jugular (harder to maintain)</td>
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<table>
<thead>
<tr>
<th>Rabbits</th>
<th>Intravenous Catheter</th>
<th>Intraosseous Catheter</th>
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<tbody>
<tr>
<td></td>
<td>Cephalic</td>
<td>Tibial crest</td>
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<td></td>
<td>Lateral saphenous</td>
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<td></td>
<td>Marginal ear vein</td>
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<td></td>
<td>Jugular (harder to maintain)</td>
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<tr>
<th>Rodents</th>
<th>Intravenous Catheter</th>
<th>Intraosseous Catheter</th>
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<tr>
<td></td>
<td>Tail (rats only)</td>
<td>Tibial crest</td>
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<tr>
<td></td>
<td>Cephalic (larger rodents)</td>
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<td></td>
<td>Lateral saphenous (larger rodents)</td>
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**Intraosseous catheter placement**

IO catheters are painful and should be placed only with an analgesic on board and possibly under general anesthesia; IV catheters are the first choice for venous access.

Aseptically prepare site. If possible inject a local anesthetic over the incision site. Small gauge spinal needles—20g, 22g, 25g—or equal gauge hypodermic needles are most commonly used. The appropriate bone is firmly stabilized and the needle placed in the direction of the long axis. The needle is rotated and pushed with steady pressure until it passes into the cavity. The needle should be firmly in place and the bone able to be moved by the needle. The catheter should flush easily. Absolute confirmation of placement can only be obtained through radiographs. Intravenous sets and infusion sets can be seated directly on the needle. The catheter can be secured with standard adhesive tape or tegaderm.

**Fluids**

Blood volume for rabbits and ferrets is 50-60ml/kg. In rodents blood volume is variant among each species, however, it is accepted that 6-8% of body weight translates into approximate blood volume. Fluid therapy should be started if a patient has deficits greater than 5% and transfusions should be stared if a patient has lost 20% or greater of blood volume. Crystalloid solutions can be given at bolus doses of 5-15ml/kg in a 15 minute period. Hetastarch can be given at 5ml/kg boluses over 10 to 15 minutes, repeating up to 20ml/kg total. Oxyglobin increases oxygen delivery to tissues and can be given as 2ml/kg boluses up to 5ml/kg in 10 to 15 minute time frames. Blood products are rarely used for rescusitative efforts for several reasons, primary being lack of available products for each species.

**Anesthesia**

As with canine and feline patients, exotic species should be pre-medicated with a sedative and analgesic combination before any anesthetic episode. Premedication helps decrease level of stress in patients, reduces anesthetic requirements, and helps to deal with pain associated with the procedure. Almost all opioids and sedatives that are used for canine and feline patients can be used in exotic patients. While it is not typically acceptable to mask induce patients, exotic species are the exception. They are usually not sedate
enough from premedication to allow for easy catheter placement. Even if IV induction is an option, many species are require more time to intubate and an induction with an IV agent may not allow enough time for intubation before it reaches a crisis state.

Rabbits are one of the more technically challenging animals to intubate; however attempts should always be made to intubate, and with practice it becomes much easier. Rabbits are intubated either nasally or using a blind oral technique. Endotracheal tube sizes range from 2.0-5.0mm. Lidocaine dripped into the nose or sprayed in the back of the mouth can help facilitate intubation. Patients must be positioned in sternal with the neck hyper-extended. This places the trachea in a straight line and makes for easier intubation. The tube is places in the nose or mouth and advanced on inspiration. Confirmation is achieved through visualization of fog in the tube or placement of a capnometer. The misplaced endotracheal tube can often be felt next to the trachea.

Ferrets are technically similar to cats in terms of intubation. Tube size ranges from 2-3.5mm, with larger sizes used in males. Lidocaine spray is useful to prevent laryngeal spasms; the mouth can be held open with gauze ties or IV tubing. Positioning is in sternal with the neck extended, very similar to positioning for feline intubation.

Rats and other rodents are extremely hard to intubate and are most commonly kept on inhalant using a tight fitting mask to limit personnel exposure.

Similar monitoring techniques are used in canine and feline patients, allowing for a smaller scale. Pulse oximetry can readily be used. Probes can be placed on the feet, ears, tails, and tongues of all species as well as being placed in the rectum or esophagus.

Doppler flow detector crystals can be placed over the dorsal pedal artery in all species and can be used in conjunction with a sphygmomanometer to obtain blood pressure readings. The Doppler probe can also be place in the femoral area, carotid/thoracic inlet, or on the chest in all species and used for sound and heart rate. Be careful not to restrict respirations if taping the probe on the chest. In rabbits the Doppler can be placed on aural artery. In rodents the doppler can also be placed on the ventral tail.

Capnometry can be used on any intubated patient and even some patients on a tight fitting mask. Dead space adapters exist that can be connected to the endotracheal tube in place of the regular tube adapter. Tube obstruction is a concern in exotic species, especially when using very small endotracheal tubes. A capnograph can be the first alert to an obstruction or inadvertent extubation and helps confirm proper intubation.

Non-invasive blood pressure monitoring is an option on some animals. Good pediatric monitors usually work well on larger rabbits and ferrets and some larger rodents. Small blood pressure cuffs can be placed on the fore or hind limbs, or on the tail in ferrets and occasionally large rats.

Electrocardiography can be readily monitored in all species. The ECG gives information about rate and rhythm and can be a helpful tool to use under anesthesia. ECG patches can be placed on both forelimbs and the left hind limb in all exotic species. It is also possible to use 25 gauge needles pierced through skin with an alligator clip attached to the needle. If using alligator clips on skin (without an ECG patch) either flatten the teeth of the clip or use gauze squares to cushion the area and prevent tissue damage.

Temperature monitoring is one of the most vital monitors in exotic patients. Small species are at a high risk for hypo- and hyperthermia. Temperature can be monitored using either rectal or esophageal temperature probes.

Small exotic patients can become hypothermic very quickly and temperature should be monitored closely for several hours postoperatively. A watchful eye must be kept on the catheter. Respiratory depression can quickly lead to cardiac arrest—vigilance can catch an issue before it becomes a critical problem. Always know the reversal drug and dose needed and have an emergency drug dose chart readily available.

References available on request.