EVALUATION OF THE SURGICAL PATIENT

Exotic companion mammals, especially rabbits and ferrets commonly present for surgery exotic animal practice, and in general tolerate the stress of handling and surgery well. Some are prey species, and therefore may react to noxious stimuli differently from traditional pet species. Therefore, careful handling and observation are critical for successful outcome. Condition can change rapidly. In some cases, the patient may be examined on one day, and then represent for surgery some days/weeks later. Reweigh the patient carefully, and be sure a physical examination has been completed on that patient the same day prior to extensive handling or the administration of any pre-anesthetic medications.

PREANESTHETIC BLOOD WORK UP

All patients benefit from blood analysis performed prior to an anesthetic procedure, preferably the day of the procedure itself. Blood values from samples collected months, weeks, or even several days prior may not reflect the current condition of the patient. In the United States, practitioners treating traditional companion animals have faced lawsuits stemming from failure to recommend pre-anesthetic blood work connected with adverse outcomes. Numerous collection sites have been described for exotic mammals, and include the lateral saphenous, cephalic veins and cranial vena cava. In rabbits, the auricular vessels are often utilized. Careful restraint is essential to prevent handling injury, and in rabbits most specifically injury to the spine caused by patient struggling and kicking. Especially fractious patients benefit from sedation (see pre-anesthesia and sedation). Minimum database includes complete blood count, or at least examination of the blood film and hematocrit; and a chemistry panel including blood urea nitrogen, creatinine, total protein, albumin, glucose and enzymes. Significant abnormalities may indicate delay or modification of the surgical plan.

PREANESTHESIA AND SEDATION

Preanesthetic agents include sedatives and analgesics, and choice is largely practitioner preference. Well-recognized benefits of the administration of preanesthetics in most patients include reduced stress of induction, and lowering of the mean alveolar concentration (MAC) of inhalant agents required to achieve a surgical plane of anesthesia. Since all anesthetic agents, including isoflurane and sevoflurane, have dose-related respiratory and cardiovascular depressant effects, attempts to lower MAC using a balanced anesthetic approach should be beneficial.

Pre-anesthetic agent combinations vary and include agents such as midazolam, opioids, ketamine and sub-anesthetic dosages of medetomidine or dexmedetomidine. All surgical patients should receive a local or line incisional block using lidocaine and bupivacaine (Table 1). Protocols are modified for ill or unstable patients.

ANALGESIA

Parenteral Analgesia

Exotic companion mammals are often laboratory mammals for analgesic studies; therefore, some data is available for selected analgesics.

Analgesia via Constant Rate Infusion (CRI)

Constant rate infusion of analgesics added to crystalloid fluids have been demonstrated to allow a reduction in mean alveolar concentration (MAC) for inhalant anesthesia in many species. While clinical studies to confirm the benefit of CRI in selected exotic companion mammals using specific agents are lacking, it is likely constant rate infusion of analgesia is useful in these patients as well. An additional indication for CRI of analgesics is for non-surgical pain.

Agents selected for constant rate infusion in the author’s practice include opioids, a combination of an opioid with ketamine and fentanyl (Table 2).
to allow accurate delivery of small volumes of fluids and drugs. Dosages are calculated as volume of drug to be administered per minute or hour, along with required fluid volume. For example, crystalloids are commonly administered at 10 ml/kg/hour to surgical patients. Analgesics are calculated as amount required per hour and added to one hour’s volume of fluids.

Local Analgesia

The addition of local analgesia reduces MAC in humans and traditional pet anesthetic patients, and has been observed by the author and others to have the same benefit in exotic companion mammal patients as well. Local analgesia should be considered as part of balanced anesthesia in all surgical patients. The most commonly used local anesthetics in the authors practice are lidocaine at 1 mg/kg, and bupivacaine at 1 mg/kg. The calculated dose can be diluted with saline to allow a volume sufficient to block the area in question.

Epidural Analgesia

Epidural analgesia has been utilized by the author and many others in ferrets, rabbits, and larger guinea pigs. The technique is identical to that in traditional companion mammals, with the injection site between the last lumbar and first sacral vertebrae in most instances. Drugs used for epidural analgesia include morphine, lidocaine and bupivacaine. Epidural placement requires anesthesia, and the use of very small spinal needles or simple injection needles (27-25g). Morphine for epidural is administered at 0.10 mg/kg.

### PATIENT PREPARATION

Pre-surgical patient preparation includes the gathering and organization of all monitoring equipment, heat sources, emergency drugs and other supplies prior to induction of anesthesia.

Vascular support is recommended for all patients undergoing anesthesia, but especially for surgeries expected to last beyond 20 minutes, or for ill or unstable patients. IV catheterization is performed routinely utilizing the cephalic vein with 24-26 g catheters.

Most calm rabbits tolerate catheterization (24-26 g) with sedation and local infiltration of lidocaine. Prepare the site for catheterization. Roll the skin over the site laterally or medially, infuse lidocaine and massage to diffuse the bleb. Return the skin to normal position and proceed, taping the catheter in place.

<p>| TABLE 1 - Drug dosages used by the author for sedation/local anesthesia of exotic companion mammals |</p>
<table>
<thead>
<tr>
<th>Drug Class</th>
<th>Drug</th>
<th>Dosage (mg/kg)</th>
<th>Route</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>Benzodiazepine</td>
<td>Midazolam</td>
<td>0.25-0.50</td>
<td>IV, IM</td>
<td>Sedation, pre-anesthesia</td>
</tr>
<tr>
<td>Opioids</td>
<td>Butorphanol Rabbit/Chin/ Guinea Pig Ferret Rat Mouse Buprenorphine Hydromorphone</td>
<td>0.1-0.3 0.1-0.2 0.3-1.0 0.5-1.0 0.04-0.05 0.10</td>
<td>IV, IM</td>
<td>Short acting; drug is very sedating in ferrets; consider lower doses. Rats and mice appear to require higher dosages of opioids. Synergistic with benzodiazepines.</td>
</tr>
<tr>
<td>NMDA antagonist</td>
<td>Ketamine</td>
<td>1</td>
<td>IM</td>
<td>Used in addition to midazolam and an opioid for additional sedation</td>
</tr>
<tr>
<td>Sedative/Hypnotic</td>
<td>Etomidate agent</td>
<td>1-2</td>
<td>IV</td>
<td>Must use with benzodiazepine to prevent seizures. Short acting induction agent.</td>
</tr>
<tr>
<td>Local anesthetic</td>
<td>Lidocaine Bupivacaine</td>
<td>1-2 mg/kg 1-2 mg/kg</td>
<td>Local bloc or infusion</td>
<td>Enhances patient comfort for procedures such as phlebotomy and catheterization</td>
</tr>
</tbody>
</table>

<p>| TABLE 2 - Drug dosages used by the author for constant rate infusion of analgesics in exotic companion mammals |</p>
<table>
<thead>
<tr>
<th>Drug Class</th>
<th>Drug</th>
<th>Dosage (mg/kg/hour)</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>Opioids</td>
<td>Butorphanol</td>
<td>0.1-0.2 0.025-0.05 0.005-0.002</td>
<td>Combine with low dose ketamine.</td>
</tr>
<tr>
<td>Hydromorphone Fentanyl</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>NM DA antagonist</td>
<td>Ketamine</td>
<td>0.4-1.0</td>
<td>Combine with an opioid. Ketamine at low dosages provides analgesia. High dosages provide sedation, but not analgesia</td>
</tr>
</tbody>
</table>
ANESTHETIC INDUCTION AND MAINTENANCE

Isoflurane or sevoflurane by facemask are the most commonly described induction agents for rabbits. As mentioned above, stress of induction is reduced by the use of preanesthetic agents, and use is highly recommended. Induction should take place with the patient carefully restrained. Handling and mask placement is performed in a calm manner in order to reduce stress.

Some animals, in particular rabbits often relax soon after induction begins, only to surprise the handler with a sudden last attempt at escape (termed the “rabbit explosion”). The handler should be certain the patient is unconscious before relaxing restraint.

Intubation allows assist ventilation and resuscitation in case of respiratory arrest, and also reduces leakage of waste anesthetic gas and exposure of personnel. Intubation is similar to that in cat in the ferret. In the rabbit, tracheal intubation is performed using the blind technique, or endoscopic-guided technique (endoscope side-by-side or over-the-top). 2.5 to 3.0 mm endotracheal tubes are suitable for most rabbits. Nasal intubation has been described, but has significant drawbacks, including poor seal and higher flow rates required to maintain anesthetic levels. Intubation in smaller patients has been described but is significantly more difficult. The guinea pig, chinchilla, and prairie dog are intubated using an endoscopic technique.

Newer injectable agents have been introduced for safe induction of anesthesia in the rabbit, and include alfaxon and etomidate. Intubation is performed immediately after induction, with maintenance of anesthesia with isoflurane or sevoflurane. Full injectable induction/maintenance protocols have also been described. The most notable indication for full injectable anesthesia is for surgery of the head and/or mouth where intubation is not possible, or when the mask and tube interfere with the surgical procedure. The most commonly reported combination for this purpose is medetomidine/dexmedetomidine and ketamine, often used in conjunction with midazolam and an opioid (table 4). These higher dosages of medetomidine/dexmedetomidine and ketamine have more profound adverse cardiovascular effects and should ideally only be used in relatively healthy patients, and with fluid support and careful monitoring of blood pressure.

PATIENT SUPPORT

Standard mammalian fluid rates for uncomplicated surgical procedures are 10 ml/kg/hour. Fluids are delivered via an infusion pump, or by intermittent hand injection. A pediatric infusion syringe pump is ideal.

Thermal support is critical for all small exotic mammal patients. Reduction in core body temperature occurs within 20 minutes of induction of anesthesia; drops are associated with cardiovascular instability, poor recovery and decreased patient survival.

Common methods of thermal support include warm water blankets, forced air heaters, electric heating pads and radiant heat. Some exotic companion mammals are especially susceptible to overheating, in particular the chinchilla; thus core body temperature should be monitored with a flexible temperature probe inserted carefully into the rectum.

ANESTHETIC MONITORING

Careful patient monitoring throughout induction to recovery enhances the probability of positive outcomes. A number of monitoring devices can aid the anesthetist; however, no device can replace the skilled and attentive veterinary nurse. Devices are often chosen based on availability and personal preference. A list of monitoring parameters and devices are presented in table 5.

RECOVERY

Patients should be monitored carefully until extubated and fully recovered. It should be kept in mind that animals that have received pre-anesthetics will experience a calm recov-

<table>
<thead>
<tr>
<th>Drug</th>
<th>Dosage</th>
</tr>
</thead>
<tbody>
<tr>
<td>Medetomidine</td>
<td>10 ug/kg</td>
</tr>
<tr>
<td>Ketamine</td>
<td>7 mg/kg</td>
</tr>
<tr>
<td>Hydromorphone</td>
<td>01 mg/kg</td>
</tr>
</tbody>
</table>

* Marla Lichtenberger, personal communication 11/08

<table>
<thead>
<tr>
<th>Drug</th>
<th>Rabbit</th>
<th>Guinea Pig</th>
<th>Chinchilla</th>
<th>Hamster</th>
</tr>
</thead>
<tbody>
<tr>
<td>Medetomidine ug/kg</td>
<td>80-150</td>
<td>70</td>
<td>70</td>
<td>100</td>
</tr>
<tr>
<td>Ketamine mg/kg</td>
<td>25-30</td>
<td>20</td>
<td>20</td>
<td>30</td>
</tr>
</tbody>
</table>

Vittorio Capello, personal communication 6/06.
ery that is slower than that seen when inhalant agents are used alone. Recovery with injectable anesthetic agents is expected to be even longer. Monitor body temperature until fully recovered. Most patients should be encouraged to eat as soon as possible after anesthesia, unless the surgical procedure indicates otherwise. Herbivores that are hesitant to begin eating should be hand fed a liquid hay based support formula (Critical Care, Oxbow Animal Health, Murdoch, NE). Carnivores are offered canned dog/cat foods, or a carnivore support formula (Carnivore Care, Oxbow Animal Health, Murdoch, NE).

### TABLE 5 - Summary of anesthetic monitoring techniques used in mammal patients

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Method</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>Respiratory (rate and depth)</td>
<td>Direct visualization</td>
<td>Newer models modified for small patients</td>
</tr>
<tr>
<td>Cardiac (rate and rhythm)</td>
<td>Stethoscope, Ultrasonic Doppler, Electrocardiogram</td>
<td>Allows hands-free monitoring, requires rapid recording speed</td>
</tr>
<tr>
<td>Blood pressure (indirect, systolic)</td>
<td>Sphygmomanometer and pediatric cuff with ultrasonic doppler</td>
<td>Requires practice, more difficult in smaller rabbits</td>
</tr>
<tr>
<td>Mucous membrane color</td>
<td>Direct visualization</td>
<td>Indirect measurement of peripheral tissue perfusion</td>
</tr>
<tr>
<td>Capillary refill time</td>
<td>Digital compression of mucous membranes</td>
<td>Indirect measurement of peripheral tissue perfusion</td>
</tr>
<tr>
<td>Temperature</td>
<td>Flexible temperature probe, Infrared thermometry</td>
<td>Place carefully into rectum, early studies in mammals promising, good correlation with other methods</td>
</tr>
<tr>
<td>Oxygen saturation</td>
<td>Pulse oximeter</td>
<td>Estimates % arterial oxygen saturation of hemoglobin, reports on usefulness variable</td>
</tr>
<tr>
<td>End-tidal carbon dioxide (ETCO₂)</td>
<td>Side stream capnograph</td>
<td>Measures CO₂ in exhaled gas (estimates arterial PaCO₂), may be unreliable in smaller animals</td>
</tr>
</tbody>
</table>

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**REFERENCES AND FURTHER READINGS**


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www.exoticvetclinic.com
A common scenario for all exotic pets is chronic disease presenting as an acute onset of illness. Many exotic companion mammals fall into the category of prey species, with inherent instincts to hide illness until unable to do so. Therefore, any animal presented in acute crisis must be carefully evaluated for long-term chronic underlying illness.

Common underlying factors in diseases affecting these species are malnutrition and improper husbandry, especially in those with difficult husbandry requirements, for example, sugar gliders. All efforts at diagnosis and treatment must include careful investigation into husbandry and explicit recommendations for correction based on the most recent understanding of the needs of these species.

The principles of emergency care and stabilization are the same as those established in human and more traditional pet medicine: airway and cardiac support, control of hemorrhage, correction of underlying fluid and electrolyte abnormalities and restoration of normothermia.

Some patients require sedation, or even general anesthesia to reduce pain and stress associated with critical care procedures. Sedatives and anesthetic agents must be chosen carefully to reduce the risk of exacerbating condition or causing death.

**AIRWAY SUPPORT**

**Respiratory arrest**

Intubation is simple in exotic carnivores; however difficulty increases with rabbits, rodents, and other species. Intubation of rabbits is achieved with the blind technique, or with endoscope or laryngoscope guidance. Both techniques require significant practice and may be impractical in the emergency setting. Forced mask ventilation has been demonstrated as a viable alternative in the rabbit, and may be applicable in other species as well. A tight fitting mask is applied over the nares and oral cavity, and ventilation supplied with an anesthetic circuit or an ambu bag. A disadvantage is accumulation of air in the stomach, which can be addressed after successful return to spontaneous respiration. Emergency tracheal intubation via tracheotomy may be an option in very small animals using a standard tracheotomy approach and small endotracheal tubes. IV catheters or red rubber catheters. The author has no experience with tracheotomy for this purpose in exotic companion mammals but assumes the risk of tracheal damage and/or stricture to be high.

**Respiratory distress**

Oxygen is ideally administered in an oxygen chamber while the patient rests undisturbed. Face mask administration may produce stress and should be avoided. In many cases, anxiety contributes significantly to dyspnea. For this reason, many patients benefit from low dose sedation using midazolam with or without butorphanol at 0.1-0.2mg/kg each administered intramuscularly. The author has seen reduction of anxiety, and decreased respiratory effort in rabbits, ferrets and rats with marked respiratory distress using low dose sedation.

**CONTROL OF HEMMORRHAGE**

Blood volume of mammals is estimated at 7-10% of body weight, and it is estimated normal healthy individuals can tolerate an acute loss of approximately 10% of blood volume. Direct pressure is often the most effective means to control hemorrhage. Silver nitrate and coagulative powder products may be used for nail hemorrhage. More severe bleeding may require ligation of the compromised vessel. The use of whole blood as part of resuscitation is discussed under fluid therapy.

**TREATMENT OF FLUID DEFICITS**

**Vascular Access**

IV catheterization is relatively easy to accomplish in the ferret (cephalic veins), the rabbit (cephalic, lateral saphenous and aural veins) and larger guinea pigs (cephalic and lateral saphenous veins). The procedure is similar to that in traditional pet species, and catheter size is 24-26g.

Intravenous catheterization is difficult for species smaller than guinea pigs. However, vascular access is feasible with intraosseous access, via the tibia or femur. The author
prefers the use of standard IV needles (27-22 g), which are placed, secured with tape and fitted with a standard catheter infusion cap. Confirmation of correct placement can be assumed by stability of the catheter and failure to accumulate fluids in soft tissues, but absolute confirmation requires radiographs of the catheter in situ in two views. Small needles used as IO catheters occasionally occlude with bone or blood clots, which can be dislodged using very fine sterilized cerclage wire as a stylet, or by removing and simply replacing the catheter.

Placement of all catheters is enhanced with the use of sedation and local analgesia, as described in detail below.

**Fluid Administration**

Optimal fluid therapy is critical for treatment of hypovolemic shock, correction of dehydration, and delivery of maintenance fluids in those patients unable to take adequate fluids PO. IV or IO administration is indicated for patients that are in shock, are depressed or hypothermic, or those judged to be over 5% dehydrated. Fluid infusion is accomplished via intermittent administration using a small volume syringe, or with a pediatric infusion or syringe pump.

For more stable patients, fluids administered SQ or PO may be adequate. In either case, fluid rates and volumes are calculated as outlined below.

**Hypovolemic shock**

While little information exists on specific guidelines for treatment of hypovolemic shock in these species, information can be extrapolated from work from other species. Lichtenberger recommends intravenous infusion of warmed hypertonic saline at 3-5 ml/kg, followed by isotonic fluids (e.g. lactated ringer’s) at 10-15 ml/kg, and colloids (Hetastarch, 6%, (B Braun Medical Inc., Irvine, CA) at 5 ml/kg over 5-10 minutes. Response to therapy (normovolemia) is best judged with measurement of blood pressure, which is possible in larger species such as the ferret and rabbit, and increasingly difficult to impossible in smaller species. Measurement of indirect systolic blood pressure is accomplished with pediatric blood pressure cuffs and a Doppler vascular monitor, in with oscillometric methods. In most exotic mammals, the cuff is placed at the humerus, and the Doppler placed in a shaved area just above the ventral forelimb footpad. Several manufacturers offer blood pressure cuffs for human digits, which can be easily adapted to limbs of small exotic mammal patients.

In the patient treated for shock, fluid administration is continued as above until blood pressure measurements read above 40 mmHg. In studied species, restoration of normal blood pressure is not possible in the hypothermic patient. Therefore, aggressive external heat support (warming blankets, warmed IV or IO fluids) is initiated simultaneously until rectal body temperature reads at least 98 degrees F (see below under “restoration of normothermia”). At this point, boluses of isotonic crystalloids (10-15 mg/kg) and colloids (5 ml/kg) are continued until systolic Doppler blood pressure reads above 90 mmHg.

When blood pressure measurement is unsuccessful or impractical, practitioners must make judgment calls regarding perfusion status based on patient response to fluid therapy (mentation) and parameters such as capillary refill time, turgor of visible surface vessels, body temperature and heart rate.

**Dehydration and Maintenance**

After successful fluid resuscitation, dehydration deficits are calculated using the formula: Body weight (kg) x 1000 x % dehydration. These fluids are replaced using isotonic crystalloids over a 6-hour period in cases of acute fluid loss, and 12-24 hours in more chronic diseases loss. Additional fluids are added for maintenance at 3-4 ml/kg/hour, and in volumes to equal estimated ongoing losses (vomiting/diarrhea). In the stable patient for which vascular access was not necessary, fluids to correct dehydration and for maintenance are calculated as above and administered in 3-4 SQ boluses. Once the patient is stable and taking adequate fluids PO, fluid administration is discontinued.

**Special fluid needs**

**Whole blood**

Rough guidelines for the indication for blood transfusion are similar to those used in other species and include acute blood loss resulting in PCV below 20%, or chronic blood loss with PCV below 12-15%. Overall patient condition (bright and alert vs. pale and depressed) is also important when considering transfusion. Whole blood should not be used in the initial phase of correction of hypovolemia.

With the exception of the ferret, small exotic companion mammals are known to have distinct blood types. However, the likelihood of transfusion reaction after a single transfusion is unlikely. The risk of reaction must be weighed against the risk of withholding transfusion. Sources of blood donors include the ill pet’s housemates, or pet stores. The author keeps a list of owners willing to provide blood donors in exchange for clinic credit. Blood is collected from healthy donors under sedation with 1 ml acid citrate dextrose (ACD) per 10 ml blood, maximum 10% of blood volume based on calculated body weight. Blood is administered via IV or IO catheter. Administer fluids using a filter to remove most of the aggregated debris. Administer donor blood by slow bolus or by infusion with a syringe pump into an IV or IO catheter. In cases of massive hemorrhage, administer blood more rapidly, within minutes. Administer blood transfusions within 4 hours of collection to prevent the growth of bacteria, according to standards set by the American Association of Blood Banks.

**Hetastarch**

Hetastarch is used in the initial phases of fluid resuscitation as described above. It can also be added to IV/IO crystalloid fluid administration (0.8 ml/kg/hour) for correction of dehydration and for maintenance in patients that are known to be hypoproteinemic.
### CPCR drug dosages suggested for use in exotic companion mammals


<table>
<thead>
<tr>
<th>Drug (concentration)</th>
<th>Dose</th>
<th>mL/50 g</th>
<th>mL/100 g</th>
<th>mL/kg</th>
<th>mL/2 kg</th>
</tr>
</thead>
<tbody>
<tr>
<td>Epinephrine (1:1000) (1 mg/mL)</td>
<td>0.01 mg/kg</td>
<td>0.0005</td>
<td>0.001</td>
<td>0.01</td>
<td>0.02</td>
</tr>
<tr>
<td>Atropine^a (0.54 mg/mL)</td>
<td>0.02 mg/kg</td>
<td>0.002</td>
<td>0.004</td>
<td>0.037</td>
<td>0.074</td>
</tr>
<tr>
<td>Glycopyrrolate (0.2 mg/mL)</td>
<td>0.02 mg/kg</td>
<td>0.005</td>
<td>0.01</td>
<td>0.1</td>
<td>0.2</td>
</tr>
<tr>
<td>50% Dextrose (diluted 50% w/saline)</td>
<td>0.25 ml/kg</td>
<td>0.05</td>
<td>0.1</td>
<td>1.0</td>
<td>2.0</td>
</tr>
<tr>
<td>Calcium gluconate (100 mg/mL)</td>
<td>50 mg/kg</td>
<td>0.025</td>
<td>0.05</td>
<td>0.5</td>
<td>1.0</td>
</tr>
<tr>
<td>Doxapram (20 mg/mL)</td>
<td>2.0 mg/kg</td>
<td>0.005</td>
<td>0.01</td>
<td>0.1</td>
<td>0.2</td>
</tr>
<tr>
<td>Vasopressin (20 U/mL)</td>
<td>0.8 U/kg</td>
<td>0.002</td>
<td>0.004</td>
<td>0.04</td>
<td>0.08</td>
</tr>
<tr>
<td>External defibrillation</td>
<td>2-10 J/kg</td>
<td>n/a</td>
<td>1</td>
<td>2</td>
<td>4</td>
</tr>
<tr>
<td>Butorphanol^b (10 mg/mL)</td>
<td>0.2-0.4 mg/kg</td>
<td>0.001</td>
<td>0.002-0.004</td>
<td>0.02-0.04</td>
<td>0.04-0.08</td>
</tr>
<tr>
<td>Buprenorphine^c (0.3 mg/mL)</td>
<td>0.04 mg/kg</td>
<td>0.006</td>
<td>0.013</td>
<td>0.13</td>
<td>0.26</td>
</tr>
<tr>
<td>Naloxone^c</td>
<td>0.02 mg/kg IV</td>
<td>0.0025</td>
<td>0.005</td>
<td>0.05</td>
<td>0.1</td>
</tr>
<tr>
<td>(0.4 mg/mL)</td>
<td>0.04 mg/kg IM</td>
<td>0.005</td>
<td>0.01</td>
<td>0.1</td>
<td>0.2</td>
</tr>
<tr>
<td>Atipamezole (reversal of medetomidine)</td>
<td>Same volume as medetomidine; IM only</td>
<td>-</td>
<td>-</td>
<td>-</td>
<td>-</td>
</tr>
</tbody>
</table>

^a Atropine (onset of action 15-30 sec) is not recommended in rabbits as many possess serum atropinesterase and dose is unpredictable. Increasing the dose of atropine increases the risk of severe tachycardia and increases the risk of ventricular arrhythmias. Use glycopyrrolate (onset of action 30-45 sec) in rabbits.


### Decompensatory phase of shock (bradycardia, hypotension, hypothermia)

Slow IV or IO bolus over 10 minutes hypertonic saline 7.2-7.5% (3 mL/kg) + hetastarch (3 mL/kg)

↓ Begin external and core body temperature warming over 1-2 hr

Begin crystalloids at maintenance rate (3-4 mL/kg/hr)

↓ When patient is warmed to 98°F (36.6°C), begin correction of hypovolemia to indirect systolic blood pressure >90 mm Hg (recheck pressure after each bolus)

Repeat boluses 3-4 times until blood pressure is normal:

1. Crystalloids (Normasol, Plasmalyte, LRS) at 10 mL/kg
2. Hetastarch at 3-5 mL/kg

↓ Positive response: indirect systolic blood pressure > 90 mm Hg:

Add maintenance fluids (3-4 mL/kg/hr)

↓ Negative response: repeat as above.

↓ No response:

Check blood glucose, electrolytes, PCV and total protein, ECG

↓ If hypoglycemic:

Give 50% dextrose diluted 50:50 with saline at 0.25 mg/kg

If PCV < 20% and low total protein:

Consider whole blood transfusion

Correct abnormal cardiac function

↓ No response: Consider vasopressor at small animal dose
**Albumin**

Human and traditional pet species often benefit from administration of colloids, or in severe cases, albumin to help raise osmotic pressure, especially in patients with marked hypoalbuminemia unable to obtain nutrition per os. Administration of crystalloids alone in these patients may result in marked loss of fluids from the vascular space into the interstitial space, resulting in worsening hypovolemia, and in severe cases pulmonary or tissue edema. A new canine albumin produce has just recently been made available, but is even more expensive. Use in any exotic species is limited and anecdotal, but should be considered in extreme cases.

**Dextrose**

Hypoglycemia is encountered in ill patients. The best correction is replacement of calories and nutrients per os. Recent trends in human and veterinary critical care have leaned away from supplementation of dextrose in IV fluids, with the exception of severe insulin overdose in humans and animals, and non-responsive insulinoma in ferrets. In a severely hypoglycemic patient unable to take oral dextrose, consider an infusion of up to 5% dextrose until blood glucose is determined to be within normal range.

**RESTORATION OF NORMOTHERMIA**

Normal rectal body temperature of companion mammal species vary widely. Measurement of body temperature is not difficult in debilitated animals. The author recommends a constant read out flexible temperature probe that can be inserted rectally, and taped into position. Depending on size, probes may not be practical in smaller species. Methods for external rewarming include heating pads, warm water bags or bottles, forced air warming devices, radiant heat sources and commercial small mammal incubators. Internal rewarming can be accomplished via infusion of warmed IV fluids, which has been shown to be extremely important for the prevention of an afterdrop effect, or return of cool fluids to the body core and worsening of condition when external warming is used alone.

**SEDATION AND GENERAL ANESTHESIA FOR CRITICAL PATIENTS**

The introduction of a number of sedatives and anesthetics with wider margins of safety has greatly increased success in exotic companion mammals. It should be kept in mind that any anesthetic or sedation procedure in a critical patient should be planned carefully, with evaluation of the risk of anesthesia/sedation vs. risk of attempting a necessary but potentially stressful or painful procedure with manual restraint alone.

The author prefers sedation with midazolam with or without an opioid in critical patients. Midazolam has a relatively wide margin of safety, and often greatly facilitates diagnostic and therapeutically procedures such as venipuncture, collection of radiographs and administration of medications. While midazolam is often effective in the critical patient when used alone, effects are enhanced with the use of lower dosages of opioids, for example, 0.1-0.2 mg/kg butorphanol. Sedation is often adequate for sample collection, diagnostic imaging, and when combined with local analgesia (1-2 mg/kg lidocaine), placement of both IV and IO catheters.

General anesthesia should be avoided in the critical patient except when absolutely indicated, for example, for emergency surgical procedures. Drugs such as ketamine and medetomidine should be avoided in critical patients, especially those with cardiovascular compromise.

**REFERENCES**


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SEDATION

Anesthesia is defined by the American Society of Anesthesiologists (ASA) as a pharmacologically induced reversible state of amnesia, analgesia, loss of responsiveness, and loss of skeletal muscle reflexes, or more simply “without sensation”. In contrast, sedation is a “drug induced depression of consciousness during which patients cannot be easily aroused, but responds purposefully following repeated or painful stimulation”. The anesthetized animal achieves a surgical plane of anesthesia, while the sedated patient does not. Degree of sedation is variable and depends on multiple factors.

The advantages of sedation primarily focus on patient safety. A 2008 study comparing death rates in dogs, cats and rabbits indicated a 2.5 times higher death rate in anesthetized vs. sedated patients. While not yet scientifically demonstrated, it is likely that sedation would also be associated with decreased risk in avian species. For this reason, sedation should be considered in place of general anesthesia in situations where anesthesia is judged to be especially risky or not essential. Additionally, sedation can be an effective adjunct to physical restraint, a means to reduce stress in hospitalized patients, and potentially a method to reduce patient memory of unpleasant procedures.

The author’s focus on the use of sedation in birds is a direct result of the perception of greater anesthetic risk in these patients, especially in those that are stressed, ill, debilitated or in respiratory distress. For some patients, the dangers associated with handling are significant, and must be weighed against the risk of foregoing diagnostic testing or procedures, or the risk of general anesthesia. For these patients, sedation provides an attractive alternative.

Disadvantages of sedation can include incomplete elimination of patient movement, patient semi-awareness, and lack of complete analgesia. Other disadvantages include risks associated with use of the drugs themselves; however these can be mitigated with careful drug selection and dosing, patient selection, and vigilant monitoring.

Drugs used for sedation

A number of drugs have reported for use in avian species, including ketamine, xylazine, medetomidine, midazolam, diazepam and butorphanol, sometimes in combination with inhalant agents. Nearly all describe use of these drugs for anesthesia; however, in some cases studies demonstrate that complete anesthesia is not achieved, and results can be better described as sedation. For the purposed of this paper, discussion will focus on drugs primarily used by the author for sedation in birds, in particular midazolam and butorphanol.

Midazolam is a benzodiazepine sedative with no analgesic effects, and is used with increasing frequency as an alternative to general anesthesia for a variety of procedures in human patients. Midazolam reduces anxiety, and has been determined to produce amnesia in humans and some laboratory species. Midazolam is water-soluble and can be administered IM, IV or SQ. There is very little published information on the use of midazolam in psittacine species. Abou-Madi described the use of midazolam combined with butorphanol for premedication of birds in 2001. A single study described use for control of seizures in an orange winged Amazon parrot with an ingested lead foreign body (Riggs 2002). A number of studies describe use of midazolam in combination with other drugs for anesthesia of guinea fowl and chickens (Ajadi et al 2009, Maiti et al 2006).

Butorphanol in an opioid analgesic agent currently considered the most useful agent of its class in avian patients (Abou-Madi 2001). Recent research has focused on pharmacokinetics, and has demonstrated safety in selected psittacine species (Sanchez-Migallan Guzman D et al, 2008, Klaphake et al, 2006).

The author and others use butorphanol at 1-3 mg/kg, in combination with midazolam, at 0.25-0.5 mg/kg for sedation in avian patients. Pharmacologic data for use of midazolam alone or in combination with butorphanol in psittacine species is currently unavailable.

Sedation in psittacines

In the author’s experience, response to administration of midazolam and butorphanol for sedation is variable, and ranges from profound to barely perceptible. Onset after intramuscular injection is rapid, within 2-3 minutes. The profoundly sedated bird does not stand, but rests on the sternum with the head over the back or down. Some rest in a head down and tail up position. Respirations are generally slow and regular. In birds with respiratory distress, respira-
Establishment of Vascular Access

Diagnostic Sampling and Imaging

Respiratory distress

A number of disease conditions produce variable degrees of respiratory distress in birds. In some cases, distress is extreme, and handling is risky. Experienced practitioners have often noticed that birds in respiratory distress display improved breathing patterns under general anesthesia. Some suggest improvement is linked to provision of oxygen. However, sedated birds in respiratory distress improve without oxygen, suggesting the benefit is more likely due to reduction of anxiety. Birds in respiratory distress are placed in a gently warmed incubator with oxygen for 10-15 minutes, then given midazolam and butorphanol by IM injection and returned to the incubator. The author has not noted a single case of worsening respiratory distress in sedated birds.

Diagnostic Sampling and Imaging

While sedation decreases anxiety and struggling during radiography, it is clear complete reduction of patient movement is superior with general anesthesia. However, calm handling and patience can result in production of high quality radiographs in patients for which general anesthesia is considered excessively risky. Collection of diagnostic samples, in particular blood is easier in the calm, sedated patient.

Establishment of Vascular Access

Most birds requiring vascular access are by necessity higher risk patients. Vascular access can be accomplished with the use of sedation, plus local analgesia over the catheterization site. Topical lidocaine gel is followed with an injection of lidocaine over the site after careful movement of the skin away from the vessel of choice to avoid inadvertent penetration. For intraosseous catheterization, lidocaine is injected subcutaneously over the desired location, and into the periostium of the bone. It is important to remember that the effects of lidocaine are not instantaneous; wait ten minutes before beginning a potentially painful procedure.

ANESTHESIA

When sedation is inappropriate (painful procedures, procedures where absolutely no movement is required), anesthesia is indicated. Examples included surgery, diagnostic imaging, and treatments that might induce pain that cannot be blocked with local analgesia. Most experienced avian practitioners no longer use general anesthesia for procedures such as blood collection. If the patient is extremely agitated or stressed, sedation is a superior option to general anesthesia.

Preparation of the patient

In many cases, patient preparation is important, and may be just as important as the procedure itself. It is often difficult to judge the optimal time for anesthesia; therefore all attempts should be made to improve the status of critically ill patients. Few procedures and surgeries are immediate emergencies, so time should be spend addressing concurrent potentially dangerous issues such as hypovolemia, dehydration, anemia and infection.

Pre-anesthetic blood work

Pre-anesthetic blood work provides useful information that may help identify factors negatively impacting surgical survival; ideally these should be addressed prior to and during the procedure or surgery. In particular, these include anemia, hydration, hypoglycemia, and hypoproteinemina. Ideally, pre-anesthetic blood work would include a complete blood count (CBC), and biochemistry panel; however, the benefits of blood work must be weighed against the risks of collection in each individual patient. Factors to consider include the availability of in-house, low sample volume modalities vs. the use of out sourced testing which may require larger sample volumes and delayed results (hours to days). At the very minimum, if patient size and condition does not allow analysis of a biochemistry panel, evaluation of the blood smear, PCV and TSS can be performed on any sized patient.

Pre-surgical fasting

The purpose of fasting is to minimize ingesta in the crop and proventriculus, and to reduce the bowel “volume” during celomic surgery. The GI transit time of most psittacines is 3-5 hours dependent of consistency of ingested material. Passerines and Rhamphastids have very short transit times of usually less than an hour and should not routinely be fasted.
Fasting time therefore depends on the species of bird, overall condition, and the presence of conditions that may impact gastrointestinal motility. In general, psittacines with a normal GI transit time should be fasted 4 hours or greater. Water can be removed and crystalloid fluids administered subcutaneously at that same time, if indicated.

Timing of fasting for birds with hypomotility can be difficult; ideally at minimum, the crop should be completely empty. In these cases, care must be taken throughout surgery and handling to limit the risk of aspiration from regurgitation.

**Pre-anesthetic drugs**

The benefits and safety of “multi-modal” anesthesia have been demonstrated in humans and many veterinary patients, and may provide the same advantages for avian patients. In general, pre-anesthetic agents can provide a smooth induction, reduce the amount of general anesthesia required, and provide pre-emptive analgesia. The ability of pre-surgical butorphanol administered IV and by constant rate infusion (CRI) to reduce isoflurane mean alveolar concentration (MAC) has been demonstrated in umbrella cockatoos. Agents most commonly advocated in avian medicine include midazolam, and butorphanol, which are discussed above under “sedation”. (Table 1). However, not all avian surgeons are in agreement over which agents are ideal, or even if pre-anesthetics provide enough benefit to overcome the perceived risks (increased respiratory depression, overall poorer outcomes).

**Vascular access**

Vascular access is ideal for all surgical patients, in particular to allow surgical fluid support and for rapid administration of emergency drugs. Intravenous and intraosseous catheters are the two options utilized; each has distinct advantages and disadvantages (Table 2).

### TABLE 1 - Peri-surgical drugs commonly cited and used in avian surgical patients

<table>
<thead>
<tr>
<th>Agent</th>
<th>Dosage (mg/kg)</th>
<th>Comment</th>
</tr>
</thead>
<tbody>
<tr>
<td>Midazolam</td>
<td>0.05-0.15 IV, IO</td>
<td>Pre-anesthesia</td>
</tr>
<tr>
<td></td>
<td>0.1-0.5 IM</td>
<td>Use lower dosages when administering IV</td>
</tr>
<tr>
<td>Butorphanol</td>
<td>0.02-0.04 IV, IO</td>
<td>Pre-anesthesia, analgesia</td>
</tr>
<tr>
<td></td>
<td>1 mg/kg IM</td>
<td>Can be reversed with naloxone</td>
</tr>
<tr>
<td></td>
<td>1-3 mg/kg IM</td>
<td>Duration of action likely short</td>
</tr>
<tr>
<td></td>
<td>1 mg/kg IV with 1 mg/kg/hour as CRI throughout surgery</td>
<td></td>
</tr>
<tr>
<td>Doxapram</td>
<td>2 mg/kg IM, IV, IO</td>
<td></td>
</tr>
<tr>
<td>Atropine</td>
<td>0.2 mg/kg IM</td>
<td></td>
</tr>
<tr>
<td>Glycopyrroate</td>
<td>0.01 mg/kg IM</td>
<td></td>
</tr>
<tr>
<td>Vasopressin</td>
<td>0.08 u/kg IM, IV, IO</td>
<td></td>
</tr>
<tr>
<td>Epinephrine</td>
<td>0.01 mg/kg IM, IV, IO</td>
<td></td>
</tr>
<tr>
<td>Crystalloids</td>
<td>10-12 ml/kg/hr IV, IO</td>
<td>Surgical rate</td>
</tr>
<tr>
<td></td>
<td>3-4 ml/kg/hr IV, IO</td>
<td>Maintenance rate</td>
</tr>
<tr>
<td></td>
<td>10 ml/kg IV, IO</td>
<td>Bolus for treatment of shock</td>
</tr>
<tr>
<td>Colloids</td>
<td>5 ml/kg IV, IO</td>
<td>Bolus for treatment of shock</td>
</tr>
</tbody>
</table>

### TABLE 2 - Benefits and risks of vascular access in avian patients

<table>
<thead>
<tr>
<th>Route</th>
<th>Advantages</th>
<th>Disadvantages</th>
</tr>
</thead>
<tbody>
<tr>
<td>Intravenous (IV)</td>
<td>Less discomfort during placement in the conscious bird</td>
<td>More time consuming</td>
</tr>
<tr>
<td></td>
<td>Confirmation of correct placement is easier</td>
<td>Difficult to secure</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Risk of exsanguination if bird removes catheter during recovery</td>
</tr>
<tr>
<td>Intraosseous (IM)</td>
<td>Placement is relatively easy, and rapid</td>
<td>More discomfort during placement in the conscious bird*</td>
</tr>
<tr>
<td></td>
<td>Can be placed in even the smallest avian patient</td>
<td>May be difficult to confirm placement without radiographs</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Cannot be placed in female birds with hormonal hyperostosis</td>
</tr>
</tbody>
</table>

* Note discomfort can be greatly reduced with the use of local analgesia 10 minutes prior to placement.
The ideal fluid rate for surgical support in psittacines is unknown; however, the authors and others use 10 ml/kg/hour. Abou-Madi reports 10-12 ml/kg/hr.

**Analgesia**

Current research supports butorphanol as the most effective and safe analgesic for psittacines available thus far. However, it should be kept in mind research has focused on a limited number of apparently healthy avian species, dosages, and administration routes, and for specific applications, e.g., pain response. Reported dose ranges vary, and studies have utilized dosages from 1-5 mg/kg administered IM, IV and/or PO. Butorphanol can be administered as a part of pre-anesthesia, delivered as constant rate infusion with fluids throughout surgery and/or post surgically at recovery. It should be kept in mind the duration of action of butorphanol in psittacine species is unknown, and appears to be relatively short. A study on plasma concentrations of butorphanol in healthy Hispaniolan Amazon parrots showed a significant decrease 2 hours post intravenous administration. If butorphanol is used as a pre-anesthetic drug, and the combined presurgical and surgical period is lengthy, there may be no effective analgesia in effect at recovery.

Local anesthesia can be utilized in avian patients. Unpublished work with birds using sedation and local analgesia alone gives support to efficacy (Lennox); however, studies using local analgesics in avian surgical patients, or in psittacines in general are lacking. Lidocaine and bupivacaine at 1 mg/kg each have been used by the author for a surgical incisional block. As exact duration of action is unknown, a portion of the total calculated dose can be retained for use at the conclusion of surgery, in particular as a splash block of the closed musculature. There is also no data on the impact of incisional/splash blocks on incision healing time; however, no clinically significant delays have been noted.

**Preparation of the surgical suite**

The following check list is helpful for preparation of the surgical suite before induction of anesthesia. Thorough description of surgical instruments, equipment and suture selection are available elsewhere.

1. **Temperature**: conservation of body temperature during surgery is important. The authors have found that warming the surgical suite itself is helpful, in addition to the use of heating pads and other devices.

2. **Instruments/Equipment**: All pre-prepared and immediately available.

3. **Monitoring equipment**: 20 plus years of practice have seen an evolution in the numbers and types of monitoring devices available and practical for avian patients. Operator familiarity with the placement and operation of monitoring devices in these patients is essential, as patient size can impact ease of use and efficacy of any equipment. Perioperative and intraoperative failure of high tech equipment is common; therefore anesthetists must be able to trouble shoot rapidly, and if a particular monitoring device cannot be quickly restored, default to a lower technical monitoring technique (e.g., replacement of Doppler with a stethoscope). Surveys of experienced surgeons demonstrate a wide variety in preferred monitoring equipment; however the most commonly cited key to success is anesthetist familiarity and experience. Table 3 outlines common monitoring devices used in avian patients.

4. **Emergency drugs** (see below).

---

**TABLE 3 - Monitoring equipment used in avian surgical patients**

<table>
<thead>
<tr>
<th>Device</th>
<th>Advantages</th>
<th>Disadvantages</th>
</tr>
</thead>
<tbody>
<tr>
<td>Stethoscope</td>
<td>Reliable</td>
<td>Not &quot;hands free&quot;, subject to displacement with movement May be more difficult to detect with hypovolemia</td>
</tr>
<tr>
<td>The attentive anesthetist</td>
<td>Only device able to detect changes in respiratory depth</td>
<td></td>
</tr>
<tr>
<td>Ultrasonic Doppler</td>
<td>Extremely reliable, even in smaller patients. Allows hands-free cardiac rate monitoring</td>
<td>Movement may dislodge probe, can be challenging to secure</td>
</tr>
<tr>
<td>Mean oscillometric blood pressure (MAP)</td>
<td>Allows monitoring of pressure trends; hands-free operation</td>
<td>Requires machine able to process rapid cardiac rates; best current machines unlikely to be helpful in birds less than 100 g. Requires manual operation</td>
</tr>
<tr>
<td>Indirect blood pressure monitor</td>
<td>Allows monitoring of pressure trends</td>
<td>Studies show poor correlation with central pressures; difficult in patients smaller than 100 g.</td>
</tr>
<tr>
<td>Pulse oximeter</td>
<td>Allows monitoring of oxygen saturation; hands-free operation</td>
<td>Probes can be challenging to use in smaller patients</td>
</tr>
<tr>
<td>Capnography</td>
<td>Allows monitoring of end-tidal CO₂. Some are equipped with respiratory monitors. Efficacy questionable in some studies</td>
<td>Required intubated patient; smaller side-stream monitors may be too large in smaller patients</td>
</tr>
<tr>
<td>ECG</td>
<td>Allows monitoring of ECG and cardiac rate</td>
<td>Requires machine able to process rapid cardiac rates</td>
</tr>
<tr>
<td>Temperature probe-flexible with constant read out</td>
<td>Allows accurate monitoring of temperature trends</td>
<td>Must be placed in the crop/proventriculus which may be difficult in smaller patients</td>
</tr>
</tbody>
</table>
Anesthetic induction and maintenance

Isoflurane is the induction and maintenance drug of choice after a 25-year history of use in avian medicine. Sevoflurane is advocated as well, with reported advantages including even more rapid induction and recovery, and lack of noxious odor.

The authors are unaware of current data comparing survival rates and safety of these two inhalant agents in birds; therefore, choice appears to be based on availability and personal preference. A 1999 study comparing induction/recovery in various psittacine species indicated recovery times were not significantly different, although there was slightly less ataxia in the sevoflurane group. No pre-anesthetic agents were used in this study.

All patients are ideally intubated; however, mucus obstruction is a potential complication in smaller patients. For this reason, some surgeons prefer to only intubate larger patients, for example, conure size and larger. Tube choice depends on patient size; however, it should be kept in mind that the relative diameter of the trachea along its length is not the same in all avian species. In other words, the diameter of the trachea of Amazona species is similar throughout the length. In contrast, the diameter of the trachea of the macaw and cockatoo rapidly decreases proximally. Therefore, an endotracheal tube that appears appropriately sized based on the appearance of the glottis is too large when advanced into the distal trachea. Oversized endotracheal tubes are associated with post-intubation stenosis in multiple species, including birds. In general, advance the tube the minimal distance required for security, and keep the neck straight to prevent kinking of the trachea and pressure against the tip of the tube.

Surgical considerations

Actual surgical techniques are discussed elsewhere. The authors have compiled a number of observations that have been useful during procedures and at recovery.

1. The longer the anesthetic procedure, the deeper the plane of anesthesia, even if settings are kept the same throughout. This is likely due to recirculation of gases within the air sacs. Gradually decrease anesthetic levels over time, especially in extended (> 30-45 minute) surgeries.

2. The most useful gauge of anesthetic depth appears to be respiratory and cardiac characteristics, for example, respiratory rate and effort (depth), and cardiac rate, rhythm and intensity obtained from an audible Doppler. Trends in the force of the signal may correlate with changes in blood pressure.

3. Painful stimulation affects the level of anesthesia required. Birds appear to react more to manipulation of the skin than to manipulation of internal organs; therefore anesthetic levels must be adjusted accordingly.

4. Increases in respiratory effort may indicate occlusion of the endotracheal tube, which is common in smaller patients with tubes less than 2.5 mm ID. In these cases, extubation and reintubation with a new tube, or maintenance by facemask may be required.

5. During surgery, the cloaca will become distended with urine and/or feces after about 30-45 minutes of surgery. In some cases this appears to be linked with cardiovascular instability, likely due to increased intraabdominal pressure. Gentle expression of the cloaca and removal of contents has been observed to result in rapid correction of abnormalities such as bradycardia and shallow respirations.

6. The most dangerous period of anesthesia appears to be the recovery period (so-called “last stitch” syndrome, reported universally by avian surgeons). Birds appear to be stable while under anesthesia, but become unstable and fail to recover when anesthesia is discontinued. A number of factors have appeared to reduce this, including adequate analgesia at recovery, and fluid support.

7. In addition to the surgeon and skilled anesthetist, having a third assistant on standby is helpful to adjust failing monitoring equipment while the primary anesthetists practices continuous monitoring using manual methods (observation/stethoscope). The third assistant can also fetch additional supplies as needed.

The surgical emergency

Procedures for how to address anesthetic emergencies should be discussed and planned out well in advance of the actual emergency. All emergency drugs should be on hand with dosages pre-calculated and pre-drawn, or with immediate access to an avian weight/dosage chart (Table 1). Principles of cardiovascular resuscitation are discussed in more detail below.

Post surgical monitoring and care

Extubation

Extubation occurs when the bird is breathing well and there is evidence of some glottal tone. Swab the mouth and glottis to remove mucus prior to extubation. Birds that have not been intubated can also form mucus; therefore swabbing is important in these patients as well.

Monitoring during recovery

Provide warmth, constant monitoring, and fluid support all the way through recovery until the bird is ambulatory and alert. After surgery, fluid rates can be reduced to maintenance (3-4 ml/hour) with additions calculated for dehydration and surgical loss. In general, fluid support is discontinued when recovery is complete; at this point birds often begin to object to and investigate the catheter and fluid line.

The delayed or poor recovery

When patients do not experience a smooth, rapid recovery, every attempt should be made to elucidate a cause, if possible, and provide treatment (Table 4). It must be kept in mind that birds administered pre-anesthetic drugs may have a delayed recovery when compared to birds that have...
received inhalant gases only. This is normal, and to be expected, but must be distinguished from a medical/surgical complication.

Pain is a potential cause of apparent delayed recovery. The painful bird is often able to stand and resist handling, but is not willing to move, and may display elevated respiratory and/or cardiac rate. In contrast, the hypovolemic bird is weak and often unable to stand or offer resistance to handling.

### TABLE 4 - Trouble shooting guide for slow recoveries in avian patients

<table>
<thead>
<tr>
<th>Potential cause</th>
<th>Diagnosis</th>
<th>Treatment</th>
</tr>
</thead>
<tbody>
<tr>
<td>Administration of pre-anesthetics</td>
<td>Rule out other causes of slow recovery</td>
<td>None required; monitor carefully</td>
</tr>
<tr>
<td>Pain</td>
<td>Rule out other causes of slow recovery; note painful posture with rapid cardiac and/or respiratory rate. Consider especially if time of last dose of opioid is more than 2 hours ago</td>
<td>Administer low dose butorphanol IV or IO and observe response over next 10 minutes</td>
</tr>
<tr>
<td>Hypovolemia</td>
<td>Take indirect blood pressure, observe basilic vein turgor, evaluate CRT from vent mucosa</td>
<td>Administer crystalloids and colloids at shock rates IV or IO as indicated above</td>
</tr>
<tr>
<td>Hypoglycemia</td>
<td>Measure blood glucose with hand-held glucometer</td>
<td>Administer glucose IV, IO, or if mild, PO</td>
</tr>
<tr>
<td>Hemorrhage</td>
<td>Look for evidence of hemorrhage, see “hypovolemia” above, note mucus membrane color</td>
<td>Administer colloids and/or whole blood; decide if necessary</td>
</tr>
</tbody>
</table>

**REFERENCES**

ASA American Society of Anesthesiologists. Continuum of depth of sedation; definition of general anesthesia and levels of sedation/analgesia. ASA 2005-10-27.


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Birds typically hide signs of illness, and many conditions produce very similar clinical pictures. Common signs of illness include lethargy, anorexia, a “fluffed” appearance, increased respiratory rate or effort, and abnormal stool production. Signs of illness in reptiles are even more limited, as reptiles have a limited behavioral repertory. The most common signs of illness in reptiles are anorexia and lethargy.

**IDENTIFICATION OF THE CRITICAL PATIENT**

Staff must be trained to recognize the potentially severely ill patient over the phone, and to instruct owners to bring it in for immediate care.

Staff must also recognize the appearance of an ill bird when it arrives in the clinic, so that the bird can be immediately transferred to the hospital.

**THOROUGH HISTORY OF THE CRITICAL PATIENT**

History must be thorough and include signalment, past medical history, source of the pet, complete diet history, caging history including whether or not the pet is always supervised, and exposure to other pets. Other important information includes recent illness and/or deaths of any other pets in the household.

**PRE-HANDLING EXAMINATION: BIRDS**

It is important to observe the patient at rest prior to restraint in order to identify those critical patients who may worsen with handling.

Observe the bird at rest at a distance, and look for visual clues, such as a slight tail bob indicating dyspnea, or decreased attention to novel surroundings. With experience, one can begin to develop a “gut feeling” for those birds who are sicker than they are appear, based on the collection of subtle clues. Frequent observation of normal birds in the clinical setting helps to quickly identify birds not completely “in tune” to their surroundings.

Before beginning restraint, be mindful of indications the bird should be released at once and the examination postponed. Dr. Teresa Lightfoot developed a helpful guide she calls the “Put It Down” list: (Note in this case “Put It Down” does not mean “euthanize”):

- Pronounced dyspnea, prolonged panting or gasping for air, inability to grasp with feet, weakness, inability to bite, closing the eyes during examination, lack of normal response to stimuli and incoordination, and marked abdominal swelling. Observation of any of the above should lead the practitioner to strongly consider releasing the bird immediately and begin planning emergency stabilization.

**The Next Step:** In general, birds can be classified as follows:

- **Bright and Responsive:** birds are bright, alert, and resist handling to some degree. Patients may be quiet, but clinical condition does not appear to have worsened after handling. There is no appearance or worsening of respiratory symptoms after handling. For these patients, sample collection can most likely proceed immediately.

- **Quiet but Responsive:** birds are quiet but alert, and resist handling to a lesser degree than normal. Clinical condition and/or respiratory symptoms worsen after handling. It is clear the stress of handling is producing a detrimental effect. For this class of patients, sample collection may need to be delayed, or may proceed after administration of fluids, followed by low dose sedation (midazolam 0.25 mg/kg and butorphanol 1-2 mg/kg IM). At these dosages, these drugs are unlikely to worsen clinical disease, but are very likely to reduce stress enough to allow sample collection to proceed. While some practitioners may prefer brief general anesthesia for the same purpose, it should be kept in mind that the risk of general anesthesia is much higher than the risk of sedation in every class of patient for which survival data has been generated. Sedation is a viable alternative that has been used in the author’s practice for many years with extremely satisfying results.

- **Depressed and Minimally Responsive:** birds appear depressed and uninterested in surroundings. Birds may exhibit respiratory symptoms, or exhibit ataxia. For these patients, sample collection is delayed until after 2-4 hours of stabilization, which generally includes warmth and fluids administered subcutaneously. (Note: attempts to place IV or
IO catheters in these birds often results in stress and death. An exception is the patient who is not responding at all to handling.

**ROUTES OF FLUID ADMINISTRATION IN ILL BIRDS**

Placement of IV or IO catheters in avian patients is well described. Ideally, oral or subcutaneous fluids should be reserved for stable, standing patients that are suspected to be less than 5% dehydrated. In reality, risk of catheter placement in some patients may outweigh benefits. Therefore, it may be beneficial to administer subcutaneous fluids along with other stabilization measures prior to catheterization. Oral fluids should not be administered if there is significant risk of aspiration, or evidence of marked gastrointestinal dysfunction.

Vascular access in birds can be performed via two routes: intravenous and intraosseous; choice depends on patient size and condition, and personal preference. Catheters may sometimes be placed successfully using manual restraint and local anesthetic only in very calm or minimally responsive patients. Patients judged to be at risk due to struggling during the procedure benefit from low dose sedation (midazolam 0.25 mg/kg with or without butorphanol at 1 mg/kg) with infusion of local lidocaine at 1 mg/kg at the catheterization site. General anesthesia is seldom required for catheterization.

IV catheters are routinely placed in cockatiel and larger sized birds, using 24-26 g catheters. Choices of site include the right jugular, basilic and medial metatarsal veins. Sites other than the jugular vein are only useful for larger birds. The smallest IV catheters the authors routinely place are 25-26 g catheters into the jugular vein of a cockatiel or small conure. The medial metatarsal catheter can be secured using tape only. Basilic and jugular catheters are commonly sutured in place. No bird should be left unattended with an IV catheter in place due to the risk of fatal hemorrhage should the bird disrupt the catheter.

Intraosseous catheterization is well described in birds, and can be performed in patients as small as a finch. Sites include the proximal tibiotarsus at the tibial crest, and the distal ulna. The relatively soft bone cortex of most birds allows the use of standard injection needles as intraosseous catheters, and size in pet birds ranges from 22 to 27 g.

Correct placement is best confirmed with a radiograph is two views (single views are notoriously misleading). As the needle can pass in and out of both bone cortices, firm seating of the needle is not always indicative of success. Fluids injected into an incorrectly placed catheter often can be detected accumulating into soft tissue spaces. However, it should be kept in mind that too much “wobble” during placement may result in a large entrance point from which fluids may leak during infusion. Proper placement of an ulnar catheter may result in branching of the basilic vein during fluid administration. The IO catheter can be capped with a standard IV injection cap and and secured by taping to the limb.

Use of IO catheters in pet birds is mostly anecdotal. Studies in human patients and some animal models indicate IO vascular access can be considered equivalent to IV access in terms of onset of action of therapeutic agents, and time to establishment of peak drug levels. Recommendations for physicians include maintenance of the catheter no more than 72 hours. Complications in humans are rare (less than 1%) and include local cellulitis and infection, fracture, and leakage of administered drugs/fluids into adjacent soft tissues. The authors are unaware of a single severe complication in an avian patient after nearly 10 years of use of this technique in clinical practice. It should be noted that placement of an IO catheter in female birds with hormone-induced hyperostosis of long bones is difficult to impossible due to accumulation of mineral in the marrow space.

**FLUID TYPES AND CHOICES**

Fluid choices include crystalloids and colloids. Individual characteristics of fluids influence type and volume of fluid administered. Crystalloids include lactated Ringer’s solution, normal saline, and hypertonic saline (7.2-7.5%). Hypertonic saline rapidly draws fluid into the intravascular space from all body compartments quickly, and can be extremely useful in selected cases of hypotension. Natural colloids are blood, plasma, or albumin. Synthetic colloids include hetastarch (HES) (Hespam, Jorgensen Labs, Loveland, CO). Isotonic crystalloid solutions are commonly used together with colloids in the resuscitation phase of shock. Warm all fluids to body temperature, regardless of the route of administration. Fluids can be warmed to 100-103°F (38-39°C) without affecting composition.

Dextrose has traditionally been added to crystalloid solutions for the treatment of hypoglycemia confirmed via blood glucose measurement; however, use is currently in question (see below). Other fluid types include albumin and whole blood, and are discussed in more detail below.

**INDICATIONS FOR FLUID THERAPY**

**Shock**

Shock is defined as poor tissue perfusion from one of two mechanisms: low blood flow or unevenly distributed flow. This results in inadequate delivery of oxygen to tissues. Hypovolemic shock is caused by absolute or relative inadequate blood volume. Absolute hypovolemia occurs with actual loss of blood, for example, arterial bleeding, gastrointestinal ulcers, or coagulopathies. Relative hypovolemia is not the result of direct blood loss (hemorrhage) from the intravascular space. Examples of causes of relative hypovolemia include severe dehydration from gastrointestinal tract loss, significant loss of plasma (burns), or extensive loss of intravascular fluids into a body space such as the abdominal cavity (celomic cavity in birds). In all cases, there is decreased blood volume and venous return to the right side of the heart. This causes a reduction in return to the left
side of the heart and reduced cardiac output. The goal of correction is restoration of blood pressure, increased cardiac performance, and maximal venous return.

In humans and some animal species, three distinct phases of shock are recognized: Early or compensatory; early decompensatory; and decompensatory. While it is unclear if the mechanism of shock is identical in avian species, some similarities have been observed, and the following descriptions are useful.

**Early or Compensatory Phase**

The early or compensatory stage of shock occurs due to baroreceptor-mediated release of catecholamines. Blood pressure increases because of the increase in cardiac output and systemic vascular resistance. This stage is documented to occur in dogs with blood loss less than 20% of their total blood volume, and was observed in ducks with blood loss less than of 25-30% of blood volume. Documented clinical signs in dogs include increased heart rate, normal or increased blood pressure, and normal or increased flow (bounding pulses and capillary refill less than 1 second). In the authors’ experience, birds commonly present in this stage of shock. An increased heart rate and normal or increased blood pressure is the key indicator of compensatory shock. Volume replacement at this stage is usually associated with a good outcome.

**Early Decompensatory Phase**

This stage occurs when fluid losses continue (estimated greater than 25-30% in birds). In studied species there is a reduction in the blood flow to the kidneys, gastrointestinal tract, skin, and muscles. There is an uneven distribution of blood flow. Clinical signs may include hypothermia, cool limbs and skin, tachycardia, normal or decreased blood pressure, pale mucous membranes, prolonged capillary refill time (CRT), and mental depression. Aggressive fluid therapy using crystalloids and colloids to support blood pressure and heart rate is required in this stage.

**Decompensatory Phase**

When significant blood volume is lost, neuroendocrine responses to hypovolemia are ineffective and irreversible organ failure begins. This appears to be the final common pathway of all forms of shock in all species. It has been observed in ducks after acute loss of 60% of blood volume (it should be noted this stage occurs at loss of 40% blood volume in dogs). Clinical signs are bradycardia with low blood pressure, pale mucous membranes, prolonged capillary refill time (CRT), and mental depression. Aggressive fluid therapy using crystalloids and colloids to support blood pressure and heart rate is required in this stage.

**DEHYDRATION**

Parameters used to estimate dehydration in mammals, including decreased skin turgor and dry mucus membranes, are less useful in birds. Severely dehydrated birds are often lethargic with a sunken eye appearance. The rate of isotonic replacement fluid administration depends primarily on the rate of fluid losses and clinical status of the patient, as indicated by the physical examination and laboratory parameters. For patients that are dehydrated but clinically stable, replace interstitial fluid deficit over 12-24 hours. If the interstitial volume was lost rapidly, replace the interstitial fluid deficit rapidly (4-6 hours). Fluid requirements for dehydration are calculated as: \( \% \text{ dehydration} \times \text{kg} \times 1000 \text{ mL} = \text{fluid deficit (mL)} \).
The formula has been found extremely useful for correction of dehydration in birds.

**Maintenance**

Maintenance fluids replace ongoing losses (vomiting, diarrhea, polyuria), meet metabolic demands, and restore intracellular water balance until the patient is eating and drinking on its own. Maintenance requirements are estimated to be higher in birds due to higher metabolic rate; therefore, the authors use a maintenance rate of 3-4 mL/kg/hr. While urinary catheterization and measurement of urine can be used to objectively determine urinary output in traditional species, this is not practical in birds.

**SPECIAL FLUID CONSIDERATIONS**

**Whole Blood**

Whole blood is indicated when albumin, antithrombin, coagulation factors, platelets, or red blood cells are required. It should be kept in mind that blood products do not promote blood flow as well as acellular fluids (e.g. HES, crystalloids). Therefore, blood is rarely used for initial resuscitation unless the patient is exsanguinating or there is a coagulopathy. The density of erythrocytes impedes the ability of blood products to promote blood flow (a viscosity effect).

As in other species, continued blood loss, nonregenerative anemia with PCV 12% to 15% or below, and clotting disorders (such as rodenticide toxicity) are indicators used to determine the potential need for a whole blood transfusion. Whole blood can be administered at 10-20 mL/kg intravenously or intraosseously. Blood donors can be homologous or heterologous species. Limited studies in selected species suggests that the lifespan of red cells transfused between homologous species is greater than that between heterologous species, for example, 1 day for homologous transfusion between pigeons, but 12 hours for pigeon to red tailed hawk. The authors maintain a relationship with a local bird rescue, and keep a list of clients willing to bring in patients for blood donation in exchange for clinic credit.

Blood collection is performed in the carefully restrained or sedated healthy donor via the jugular vein, using a needle.

**TABLE 1 - Suggestions for correction of perfusion deficits in avian patients**

<table>
<thead>
<tr>
<th>Decompensatory phase of shock (bradycardia, hypotension, hypothermia)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Slow IV or IO bolus over 10 minutes hypertonic saline 7.2-7.5% (3 mL/kg) + hetastarch (3 mL/kg)</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>Begin external and core body temperature warming over 1-2 hr</td>
</tr>
<tr>
<td>Begin crystalloids at maintenance rate (3-4 mL/kg/hr)</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>When patient is warmed to 98°F (36.6°C), begin correction of hypovolemia to indirect systolic blood pressure &gt; 90 mm Hg (recheck pressure after each bolus)</td>
</tr>
<tr>
<td>Repeat boluses 3-4 times until blood pressure is normal:</td>
</tr>
<tr>
<td>1. Crystalloids (LRS, Normasol, Plasmalyte) at 10 mL/kg</td>
</tr>
<tr>
<td>2. Hetastarch at 3-5 mL/kg</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td><strong>Positive response:</strong> indirect systolic blood pressure &gt; 90 mm Hg:</td>
</tr>
<tr>
<td>Crystalloids to correct dehydration plus ongoing losses (Table 38-4)</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>Add maintenance fluids (3-4 mL/kg/hr)</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td><strong>Negative response:</strong> indirect systolic blood pressure &lt; 90 mm Hg:</td>
</tr>
<tr>
<td>Repeat as above</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>No response:</td>
</tr>
<tr>
<td>Check blood glucose, electrolytes, PCV and total protein, ECG</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>If hypoglycemic:</td>
</tr>
<tr>
<td>Give 5% dextrose IV</td>
</tr>
<tr>
<td>If PCV &lt; 20% and low total protein:</td>
</tr>
<tr>
<td>Consider whole blood transfusion</td>
</tr>
<tr>
<td>If abnormal cardiac function:</td>
</tr>
<tr>
<td>Correct contractility (nitroglycerin 1/8 inch/2.5 kg)</td>
</tr>
<tr>
<td>↓</td>
</tr>
<tr>
<td>No response:</td>
</tr>
<tr>
<td>Consider vasopressor at small animal dose</td>
</tr>
</tbody>
</table>

Reprinted in part from the conference of the Association of Avian Veterinarians, 2011.
and syringe, or butterfly catheter. Collect blood into syringes with sodium citrate, at product recommended doses. The blood-administration set must include a filter to remove most of the aggregated debris. Administer donor blood by slow bolus or by infusion with a syringe pump into an IV or IO catheter. In cases of massive hemorrhage, administer blood more rapidly, within minutes. Administer blood transfusions within 4 hours of collection to prevent the growth of bacteria, according to standards set by the American Association of Blood Banks.

**Albumin**

Human and traditional pet species often benefit from administration of colloids, or in severe cases, albumin to help raise osmotic pressure, especially in patients with marked hypoalbuminemia unable to obtain nutrition per os. Administration of crystalloids alone in these patients may result in marked loss of fluids from the vascular space into the interstitial space, resulting in worsening hypovolemia, and in severe cases pulmonary or tissue edema. Human albumin has been used successfully by one author (Lichtenberger) for treatment of severe hypoalbuminemia in a bird with chronic regurgitation and diarrhea. Human albumin is relatively expensive; last price check was approximately $85.00 for 50 ml.

This product cannot be saved after opening. A new canine albumin product has just recently been made available, but is even more expensive. Application for use in birds is currently unknown.

**Dextrose**

Hypoglycemia is encountered in ill birds. The best correction is replacement of calories and nutrients per os. Recent trends in human and veterinary critical care have leaned away from supplementation of dextrose in IV fluids, with the exception of severe insulin overdose in humans and animals, and non-responsive insulinoma in ferrets. In a severely hypoglycemic bird unable to take oral dextrose, consider an infusion of up to 5% dextrose until blood glucose is determined to be within normal range.

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Anesthesia in reptiles is well described in terms of anecdotal use in clinical practice; however there are few scientific studies for use in this extremely diverse group of patients. Complicating a uniform approach to anesthesia is markedly variable resting metabolic rate among species, and the effects of environmental temperature on metabolism, among other factors. A 2005 survey of reptile veterinarians showed that the most commonly utilized agents for anesthesia, sedation and/or analgesia were isoflurane, ketamine, propofol and butorphanol. These veterinarians agree that anesthesia is challenging, and respiratory depression, difficulty in measuring anesthetic depth, prolonged recovery and hypothermia were listed as the most common complications.

A thorough discussion of reptilian physiology as it pertains to administration of anesthesia is beyond the scope of this paper. A number of points are helpful when considering anesthesia in reptiles:

1. Reptilian resting metabolic rate is lower than that of similarly sized mammals, and is markedly variable among reptilian species.
2. Reptile metabolism is highly dependant on environmental temperature. Therefore, environmental conditions influence anesthetic time of onset and recovery, and duration of effect.
3. Reptiles are considered “episodic breathers”, and breath holding complicates administration of inhalant induction agents.
4. Ventilation (respiratory rate and tidal volume) is reduced in an oxygen-rich environment.

### PRE-ANESTHETIC FASTING

Timing of pre-anesthetic fasting is difficult in reptiles, as so many factors influence gastrointestinal transit time. In general, avoid feeding the reptile 1-2 days prior to surgery.

### ROUTES OF ADMINISTRATION

Anesthetic and analgesic agents can be administered by routes commonly utilized in other species, including IM, IV or IO, and via the respiratory tract. Use of drugs designed for intravenous administration depends on the ability of the practitioner to competently perform venipuncture or secure vascular access. Even experienced reptile practitioners report IV access as challenging, and in many cases, stressful for the patient. Access sites for IV anesthetic administration depends on the anatomy of the species in question, and include the ventral coccygeal vein of snakes and lizards, jugular and coccygeal vein in tortoises, and abdominal vein of lizards. Intramuscular administration is common, and most practitioners avoid the use of the hindlimb musculature to avoid the first pass effect produced by the renal portal system. Although recent studies on portal circulation in selected reptile species downplay this feature, it is probably best to avoid administration of nephrotoxic drugs or those highly metabolized by the kidneys into the rear limb or epaxial musculature. Studies in humans and other mammals have shown that intravenous administration of drugs is nearly identical to intravenous administration. A study examining renal function in the green iguana showed similar uptake when radioactive substances were administered IV or IO.

Mask or chamber induction with inhalant agents is commonly described, but often requires extended induction periods due to breath holding.

### PRE-MEDICATION

In traditional pet species, premedication is recommended to provide analgesia, ameliorate the stress of anesthetic induction, and to reduce the dosage and thus potential untoward effects of any single agent. These benefits can be...
expected in reptiles as well, and premedication may allow quicker induction, and permit procedures such as catheterization and endotracheal intubation. Large, powerful or venomous species may require preanesthetic drug administration to facilitate safe handling and anesthetic induction. Unfortunately, clinical trials on the use of these drugs in various reptile species are lacking and reports suggests results are highly variable.

The most commonly reported preanesthetic agent used in reptiles is butorphanol. In other species, administration of butorphanol often results in reduction of isoflurane mean alveolar concentration (MAC). However, a single study in the green iguana showed that butorphanol had no such effect.6

Others report use of midazolam in combination with an opioid agent like butorphanol or buprenorphine. The author and others often find this combination will reduce patient struggling during manipulation and induction somewhat, but results are highly variable.

Acepromazine is not considered a useful drug for preanesthesia of reptiles.2 Other injectable agents can be used for preanesthesia in preparation for induction of anesthesia, and include ketamine, xylazine, medetomidine and combinations such as tiletamine and zolazepam.2 Higher doses of these drugs often result in prolonged recovery times.

There is no conclusive data on the efficacy of analgesics in reptilian species. However, most agree the use of analgesia is a humane choice, especially when considering potentially painful procedures.2

**INDUCTION OF ANESTHESIA**

The most commonly reported induction agent in use in reptiles is propofol, as results tend to be reliable and predictable.1,2 However, use depends on the ability of the practitioner to achieve IV access, which can be difficult and stressful. Other injectable induction agents typically used include ketamine, xylazine, medetomidine and combinations using dexmedetomidine, again with variable results described from good to poor. Success and safety are often enhanced when these drugs are used in combination with preanesthetic agents, as described above.2

Simple mask or chamber induction with inhalant agents can be prolonged, as long as 13 minutes in Dumeir’s monitors.3 Alternatively, reptiles can be restrained and intubated while conscious, and then manually ventilated to achieve anesthesia. This technique is expected to produce stress, and should be utilized only when necessary.2

**ALFAXALONE IN REPTILES**

Recently, much attention has been focused on the use of alfaxalone (Alfaxan, Jurox, NSW, Australia), which is currently the author’s drug of choice for induction of reptile patients. Alfaxalone is an injectable anesthetic agent used for induction and maintenance of anesthesia in dogs and cats. The drug is available in Australia, and the UK, but not currently manufactured and distributed in the United States.

In dogs and cats, the drug can be used as a repeated bolus, or as a constant rate infusion (CRI) as part of total intravenous anesthesia (TIVA). As a CRI, the drug does not accumulate and recovery does not appear prolonged. This is supported by the observation that repeated bolus doses in reptiles do not appear to prolong recovery.

Alfaxan is frequently used in dogs and cats with premedications, including benzodiazepines and opiates. It is described for slow intravenous use, with a recommended rate of administration of about 60 seconds. Alfaxan is labeled for intravenous use only. However the drug causes no irritation or untoward effects if administered extravascularly. The manufacturer does not recommend combining Alfaxan with other drugs into the same syringe.

Alfaxan appears to have a wide margin of safety. In a study in dogs, patients were administered a 10 times overdose and survived, but required respiratory support. Cats survived a similarly at a 5 times overdose.

**Use of alfaxalone in reptiles in clinical practice**

The author and others have experience with the use of Alfaxan in reptiles in clinical practice. The motivation for use of Alfaxan are two fold: frustration with the array of drugs currently recommend for IM use in reptiles available in the United States, and technical difficulties associated with reliable intravenous administration.

Some reptile practitioners claim near 100% success rate with intravenous administration of anesthetics such as propofol in reptile patients. Skepticism aside, for those in practice with lesser talent, an effective, consistent intramuscular anesthetic agent with a reasonable recovery time is attractive.

The best uses for Alfaxan in reptiles appear to be the following: a) induction (with or without pre-anesthetics) followed by immediate intubation and maintenance with isoflurane; and b) sedation (with or without other agents) combined with local analgesia for brief, minor procedures. Even when combined with pre-medications, Alfaxan alone does not appear to achieve an acceptable surgical plane of anesthesia at currently explored dosages. Duration of action is variable but in general brief, often no more than 15 minutes. Full recovery is usually within one hour, but can be longer when combined with other agents, including inhalant agents, and especially in debilitated patients.

Dosages ranges based on the author’s experience are 5-25 mg/kg. Ill or debilitated patients require significantly less drug than fractious, more stable patients. Dosages required appear to be higher in chelonians and green iguanas, and lower in snakes and leopard geckos. The author always begins with the lower end of the dosage range (5-10 mg/kg), adding boluses as needed to effect.

The author has experienced only one fatality directly related to administration of Alfaxan, in a moderately debilitated green tree frog with a rectal prolapse. The author also was unable to inject enough Alfaxan (before running out) to achieve anything close to sedation in a 25-pound sulcatta tortoise.
MAINTENANCE OF ANESTHESIA

Inhalant agents (isoflurane, sevoflurane) are overwhelmingly preferred for maintenance of anesthesia, and intubation is practical in all but the smallest reptile species. The glottis is easily visualized, and the availability of medium to very small sized endotracheal tubes facilitates intubation.

ANALGESIA

As discussed above, very little is understood about effective analgesia in reptiles. The author combines an analgesic in pre-medication (usually butorphanol) with a lidocaine/bupivacaine incisional block at 1 mg/kg each combined and diluted with saline to desired volume. Toxic doses are unknown, but the author has found no complications with the use of these dosages.

MONITORING OF ANESTHESIA AND PATIENT SUPPORT

Most reptile patients do not spontaneously breath while under general anesthesia; therefore patients must be periodically mechanically ventilated (1-2 breaths per minute), or ventilated with a mechanical ventilator.

Retention of body heat is critical, and temperature should be monitored with a flexible constant read out temperature probe inserted into the oral cavity and into the esophagus. Body heat is maintained with active warming of the patient, and of administered fluids.

Vascular access can be difficult, but choices include IV catheter placement (often achieved with a cut down of the jugular vein), or IO placement in reptiles with limbs. Surgical fluid rate has not been established for reptiles, but is assumed to be lower than that of birds and mammals. The author used 1-2 ml/kg/hour.

The best cardiac monitor for reptiles is the ultrasonic Doppler, which is placed as close to the heart of the patient as possible, and taped into place.

Measurement of blood pressure is difficult in reptiles, and seldom used.

THE SURGICAL/ANESTHETIC EMERGENCY

Procedures for how to address anesthetic emergencies should be discussed and planned out well in advance of the actual emergency.

All emergency drugs should be on hand with dosages pre-calculated and pre-drawn, or with immediate access to a weight/dosage chart (Table 1).

Very little is reported on the use of emergency drugs in reptile patients. Anecdotally, most practitioners extrapolate from known mammal/avian drugs with variable success.

POST SURGICAL MONITORING AND CARE

Recoveries in reptile patients are usually long, and time from end of anesthesia to purposeful movement may be many hours.

Ventilation should continue with the use of an ambu bag, (not with pure oxygen, as respirations are inhibited in an oxygen rich environment) until the patient begins spontaneous movement.

Recovery will be prolonged if the patient is below optimum body temperature.

TABLE 1 - Injectable agents preferred by the author for anesthesia and analgesia of most common reptiles

<table>
<thead>
<tr>
<th>Drug</th>
<th>Dosage (mg/kg)</th>
<th>Primary Usage</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>Butorphanol</td>
<td>0.4-2.0 SQ, IM IV</td>
<td>Pre-anesthesia and analgesia</td>
<td>Efficacy uncertain in many species</td>
</tr>
<tr>
<td>Buprenorphine</td>
<td>0.02-0.2 mg/kg SQ</td>
<td>analgesia</td>
<td>Efficacy uncertain in many species</td>
</tr>
<tr>
<td>Morphine</td>
<td>0.05-1.0 mg/kg IM, IC, SQ</td>
<td>analgesia</td>
<td>Efficacy uncertain in many species</td>
</tr>
<tr>
<td>Midazolam</td>
<td>1-2 IM, IV in all species</td>
<td>Pre-anesthesia</td>
<td>Used in combination with butorphanol</td>
</tr>
<tr>
<td>Alfaxalone</td>
<td>5-25 mg/kg IM</td>
<td>Induction or sedation</td>
<td>Author’s drug of choice, combined with butorphanol</td>
</tr>
<tr>
<td>Ketamine</td>
<td>5-20 IM lizards 10-60 IM snakes 5-50 IM chelonians</td>
<td>Pre-anesthesia and anesthesia</td>
<td>Results extremely variable, use in combination with other pre-anesthetics to reduce required amount</td>
</tr>
<tr>
<td>Propofol</td>
<td>3-5 IV, IO</td>
<td>Induction of anesthesia</td>
<td>Induction agent of choice; requires intravenous or intraosseous administration</td>
</tr>
<tr>
<td>Isoflurane</td>
<td>Induce at 5%, maintain at lowest effective concentration</td>
<td>Induction and maintenance of anesthesia</td>
<td>Not recommended as sole agent due to high concentrations and time needed for induction; use in combination with pre-anesthetic and induction agents</td>
</tr>
</tbody>
</table>
REFERENCES


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