Safe and as stress free as possible animal handling, physical exams for nursing purposes and blood collection are fundamental skills every veterinary technician must know. From exams and blood samples, we gather pivotal information regarding a patient’s health. As technicians we are often challenged with working with a variety of species from large to tiny and are required to be proficient no matter what species or size. Physical exams mimic those performed in canine and feline species with careful attention to the auscultation of the lungs, heart and palpation of the abdomen, lymph nodes and peripheral limbs. A detailed history of an animal husbandry and normal habits is crucial in patient assessment of any species. However, for some species there are particular anatomical and physiological differences we must observe. Ectotherms, more commonly know as “cold blooded” animals will have a variable temperature or may not acquire a fever like in endothermic species. Birds and reptiles also have air sacs instead of lungs like mammals, birds also have sinuses, making auscultation altered compared to mammals. Listening for clear inspiratory sounds is all may one will be able to assess in animals with air sacs for their respiratory system. One may not be able to hear anything at all in some species.

Sample collection is particularly intriguing in exotic species. Thankfully our technologies are advancing, and samples required for diagnostic purposes have dramatically shrunk; yet the quality of the sample is important in meeting the minimized volume requirements.

Sample size is of the upmost importance when it comes to patient safety. The general guideline for total volume collected among all species is no more than 10% of the patient’s total blood volume. To be more specific in situations of multiple blood collection, blood volumes should be kept within the 10% volume limit until the blood volumes can self-regenerate. In mammals total blood volumes are around 6-8% of their body weight, avian and reptile species 5-8%.

Below are two charts describing the consequences of blood volumes collected and the regenerative times for differing volumes collected in mammals. These numbers are similar in avian and reptile species, but can be increased slightly with supportive care. Reptile species however can have an extremely prolonged circulating blood recovery time as their erythrocytes last anywhere from 600-800 days.


Other concerns related to blood collection include hemorrhage with associated bruising, lacerating the vessel and injury to surrounding tissues, infection and possibly hypotension depending on the volume collected. When larger volumes of blood are collected replacing the volume taken with crystalloid fluids at the same volume or even slightly greater are recommended to prevent hypovolemic syndromes.
Appropriate supplies for blood collection include a wide variety of needle, butterfly catheters, micro-hematocrit tubes and syringe sizes chosen accordingly to vessel size and location. Choosing the appropriate syringe with limited pull back is important to prevent vessel collapse and hemolysis of samples. After collecting the sample having the appropriate tube with anti-clotting agents or blood preservatives at the correct volume ratio is needed to prevent hemodilution or skewed results.

Restraint if the patient is not sedated will be crucial for successful blood collection as well as patient safety. Handling resources for different species can be found at the following websites.

http://www.ahc.umn.edu/rar/handling.html

http://web.jhu.edu/animalcare/procedures/restraint.html

Sedation/anesthesia is by far the safest method for reducing trauma associated with various blood collection techniques or physical exam of wild or highly stress patients, with an assumed risk with the sedation or anesthesia. Whenever possible, using medications that can be reversed is ideal or use sedatives that can be metabolized quickly. Using a combination of sedatives/anesthetics can help alleviate large doses of a single drug and potentially reduce negative cardiovascular and respiratory effects when particular medications are used. The other benefit of a multimodal approach is a shorten recovery time, again compared to using larger single medication doses.

Collection sites will vary among species and the anatomy of an animal. Below is a chart of common sites for blood collection in mammal species.

<table>
<thead>
<tr>
<th>Species</th>
<th>Method</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>Ferret</td>
<td>Cranial vena cava</td>
<td>Approach especially easy and safe due to particular anatomy of the ferret, even with manual restraint</td>
</tr>
<tr>
<td></td>
<td>Jugular vein</td>
<td></td>
</tr>
<tr>
<td></td>
<td>Tail vein</td>
<td></td>
</tr>
<tr>
<td>Rabbit</td>
<td>Auricular or central ear artery</td>
<td>Reported risk for vessel damage with auricular method, rare in author’s experience</td>
</tr>
<tr>
<td></td>
<td>Lateral saphenous vein</td>
<td>Restraint relatively easy</td>
</tr>
<tr>
<td></td>
<td>Medial femoral vein</td>
<td>Restraint relatively easy</td>
</tr>
<tr>
<td></td>
<td>Cranial vena cava</td>
<td>Manual restraint for vena cava approach extremely stressful and not recommended</td>
</tr>
<tr>
<td></td>
<td>Cephalic vein</td>
<td>Reduced size makes adequate sample collection difficult</td>
</tr>
<tr>
<td>Guinea pig</td>
<td>Lateral saphenous vein</td>
<td>Restraint relatively easy, is located more proximally than expected; shaving enhances visualization</td>
</tr>
<tr>
<td></td>
<td>Cranial vena cava</td>
<td>Manual restraint for vena cava approach extremely stressful and not recommended</td>
</tr>
<tr>
<td></td>
<td>Cephalic vein</td>
<td>Reduced size makes adequate sample collection difficult</td>
</tr>
<tr>
<td>Rodents</td>
<td>Cranial vena cava</td>
<td>Manual restraint for collection from any site is extremely difficult due to the small size of these patients</td>
</tr>
<tr>
<td></td>
<td>Lateral saphenous vein</td>
<td></td>
</tr>
<tr>
<td></td>
<td>Tarsal vein</td>
<td></td>
</tr>
<tr>
<td></td>
<td>Lateral tail vein</td>
<td></td>
</tr>
<tr>
<td>Hedgehogs, gliders</td>
<td>Cranial vena cava</td>
<td>Manual restraint impossible in all but extremely debilitated patients</td>
</tr>
</tbody>
</table>

Other blood collection sites include the following:

Submandibular bleeding for mice or other small rodents is a relatively simple way to obtain larger sample sizes quickly, but does require sedation or anesthesia. The technique requires the technician to puncture the area behind the ramus of the mandible and maxilla. Veins that drain blood from other parts of the face meet in this area and form the jugular vein. Once sedate one must scruff the mouse and poke a small hole. Various items may be used to puncture the skin including 19, 22, or 23 gauge needles, number 11 scalpel blades, or specially made lancets. Once a punctured the site should begin to bleed rapidly and the drops can be caught in a sample collection tube. Pressure is then applied to the puncture site until the bleeding stops.

Retro-orbital bleeding techniques are falling out of favor in many labs do to animal welfare concerns and is impractical in the clinical setting. However the technique still is a good way to collect larger samples. This technique requires heavier sedation/anesthesia and a skilled technician familiar with the technique. The mouse is first scruffed, then using a micro hematocrit tube the technician will, with a quick and semi forceful manor, twist the tube under the eyeball near the lacrimal caruncle until in the retro orbital sinus. One will know if they were successful by blood flow or not. Immediately after the procedure any residual blood should be cleaned and a copious amount of ophthalmic lubricant placed to prevent corneal trauma. If the eyeball appears bulged, blood shot, cloudy or squinted for long periods of time post procedure may indicate trauma.

Tail vein or tail arterial blood collection is also a common site for smaller sample sizes and is commonly performed in rats and mice. Typically a 27g or 30g needle will be needed to access these smaller vessels.

A cardiac stick is for anesthetized and terminal animals only! This method allows for the largest collection sample.

Tail nipping or cutting off a toe of any animal is now deemed inappropriate for animal welfare concerns and should be avoided in chronic patients.

In avian species blood collection is most easily attained from the jugular, basilic (ulnar) vein or medial metatarsal veins.

Reptile and amphibian blood collection is particularly tricky as most blood draws are done blindly. In snakes the ventral coccygeal vein is used for mid to larger sized specimens. Cardiocentesis is appropriate for any size of snake, frogs and toads but may be ideal for almost any smaller animal.

Lizards and Salamanders: Similar to snakes, blood can be collected from the ventral coccygeal vein. The mid abdominal vein is also a good site in animals that can be restrained easily. Accidental laceration of this vessel can lead to intra-coelomic hemorrhage. The jugular vein is a vessel that can be accessed depending on species anatomy, located approximately midline on the lateral aspect of the neck.

Chelonians: Blood collection is easiest under sedation or anesthesia to prevent the donor retracting into their shell. Sites easiest for collection include the jugular vein, subcarapacial sinus and dorsal coccygeal sinus. Other sites include the brachial or cephalic vein, but can be difficult in smaller species of chelonians.
Enter the links below for a video on blood collection from a snake and fish:

(Snake) [https://www.youtube.com/watch?v=5RluUKv5Asw](https://www.youtube.com/watch?v=5RluUKv5Asw)

(Fish) [https://www.youtube.com/watch?v=9kpxB5bnC7I](https://www.youtube.com/watch?v=9kpxB5bnC7I)

In the past, pain management among all veterinary species was often looked over or thought unnecessary. It was also thought that pain and associated sedentary behavior would deter the patient attention to the surgical site. Or, even worse, that some species do not feel pain. We all know now this is a myth. Most, if not all, sources now refer to a more anthropomorphizing approach. What this means is if the procedure or condition the animal is going through would be painful to you, it is undoubtedly painful to your patient. Many of the drugs we use in exotic species and the doses that we use them at are based on either pharmacokinetic studies, extrapolations from other species or from more invasive tests and assays. These invasive tests and assays are usually tested on common laboratory species in a research setting and are described below. With pain management, it is often better to slightly overestimate the pain management protocol than to underestimate our protocols as pain can not only be uncomfortable, but also have more serious physiologic and psychological effects.

Despite studies with nociceptive tests and generalized pain assessments for certain species, we still lack in veterinary medicine a well-validated generalized pain scoring system. A recent article published in 2013 sums up a culmination of studies that show fMRI can differentiate pain in the brain for humans. Although these studies were performed on human subjects, it does open the door for further research into pain detection and stimuli in animal models.

Like everything in medicine, our pain scoring systems are ever changing and being refined as more scientific and behavioral data is published. Our advocacy for the animals in our care is as vital as any other aspect of our job. In human hospitals there are such positions as “Patient Advocates” that we as veterinary nursing staff should modify to accommodate our patients as well. Working in the front lines of patient care, no one other than the patient’s owner or keeper will have spent more time and know the animal as intimately as you. Pain management protocols should be thoroughly thought out and designed the same way as in our small animal patients. Protocols should include pre-emptive analgesia to alleviate “wind up”, multimodal analgesia to cover central and peripheral pain receptors, intra-operative and post-operative management during noxious stimuli or greater stimulations of pain. Many times exotic animals must be anesthetized for physical exams. With the additional of analgesia and its MAC sparing abilities, appropriate pain management or lock can make anesthesia safer, by reducing the percentage of volatile anesthetics the patient would normally receive without analgesic adjuncts.

Many of the same localized pain management techniques used in small animals (i.e. splash blocks, nerve blocks, infusion catheters, epidurals) can be adapted for exotic species, which can greatly reduce central sensitization. However, it is imperative to calculate doses of blocking agents as to not unintentionally overdose the species we are working with.) Thorough individual research should be conducted prior to using local anesthetic methods as well. Careful consideration must be paid as to where the animal species falls in the evolutionary development chain; as suspected orders of receptor evolution follow a distinct path starting with the kappa receptor then delta and most recently evolved mu opioid receptors. There is always new research underway changing older, formerly proven methods that may not be recommended any longer, being found as unsafe or ineffective.

**Analgesia in the Avian Species**
Pain in birds serves its purpose the same way it does in all species, to provoke a response by the animal to remove itself from the stimulus. Avian pain pathophysiology is slightly different from mammals. Although largely similar to mammals, the main difference comes from the organization of the forebrain of bird’s verses mammals and the ascending nociceptive system. Birds have more kappa receptors in general. However, among bird species there are varied amounts of kappa opioid receptors located in the forebrain, yet still higher than mu opioid receptors in the forebrain in all species. This explains the varied response to mu agonists compared to mammals. It is now accepted birds perceive pain similarly to that in mammals, still having many of the same nociceptive receptors. As prey species birds are masters of pain disguise and will have one of two responses to pain, a “fight and flight” response or “Withdrawal/conservation” response.

Bird experts have thought that chronic pain is more associated with the “Withdrawal/conservation” responses, commonly seen in practice compared to the “fight or flight” response associated with sudden and unexpected pain stimuli. Birds, like other species, have adrenal stimulation under stress with a subsequent release of corticosterone, which has an adverse effect on both the immune system and wound healing. A bird’s stress level is even more exaggerated when in a small cage and its inability to feel that it can fly or get away. Therefore, it is imperative to keep the patient in a quiet, light-regulated environment far away from visible predatory species or other stressors.

Many pharmacokinetics studies have been performed on a variety of different species of birds. Summing all the studies up we see a varied and diverse array of efficacy in the varying species of birds. Where one drug like, butorphanol, was found to have analgesic effects on African grey parrots, but did not in blue-fronted Amazon parrots. There are numerous examples of this kind of outcome in the literature today. This kind of research shows not only further research needs to be performed, but also as veterinary professionals in the frontlines we must make careful and accurate assessments of our patients, starting with the physical exam. If possible the physical exam should be performed with as little stress as possible to get a set of reasonably normal baseline vital signs and behavior patterns. This may include leaving the bird alone and watch from afar.

Drug delivery in birds, like most exotic species, can be tricky. While oral dosing is easiest birds often will need to be gavaged, or syringe fed medication with restraint. This can easily stress the bird and lead to aspiration. If the bird is going to be manually restrained anyway, one might consider injectable medications instead to reduce aspiration risk and confirm adequate dosing. Common sites for intramuscular injection on a bird are the pectoral muscles and if large enough the thigh muscles. It is important while restraining a bird that it is done properly. Birds use their whole body to breath and squeezing a bird may inhibit normal tidal volume intake. If the bird is going to be anesthetized or is easily handled intra-venous access is also an acceptable method, however IV injections often wear off quicker. Subcutaneous injections are often given in the inguinal region of birds.

Analgesia in Reptiles

Reptile pain management protocols should be thought of and implemented the same way as any other species. Within varying reptilian orders are huge physiologic and behavioral adaptations making pain assessment subjective. If there is a question as to whether a procedure or condition of the patient warrants analgesia, the patient should receive the benefit of analgesia.

A physical exam with baseline vitals should be conducted. One may have to step away from the animal to observe normal non-stressed respiratory rates as with birds. In some species of reptiles, like snakes, one can even see the heart beat through the skin.

Often, if not all of the time, reptiles exhibit no obvious signs of pain other than changes in behavior. Therefore, to be effective pain management advocates it is imperative to know what a particular species and patient’s behavior patterns are. This is where detailed and accurate client communication is vital as well as an excellent patient history.
The exact pathophysiology of nociception in reptiles is still not greatly understood. However, neuroanatomical components for nociception have been described in multiple reptile models. This is strengthened by known pharmacologic agents that provide analgesic effects in other species providing similar to the same affect in some reptile species. Spinal projections from the brainstem have been found to tract mediation of descending inhibition of nociception in one species, the Tokay lizard gecko. Similar neurotransmitting features to that in mammal nociceptive modulation have also been described. Red-eared sliders have both functional mu and delta receptors in their brains, while at least 2 species of snakes have endogenous brain opiates. This pathology, as with many reptile experts, clearly suggests that reptiles do feel pain similarly to that in mammals. Even with this evidence the full understanding of reptile nociception is unclear in regards to “perception”.

Not only do reptiles have an interesting and variable neuroanatomical and receptor pathology, but a varying and tricky systemic use of drugs in general. A study in red-eared sliders discovered that the femoral veins drained directly into the abdominal vein, which then went straight into the liver. The liver being a filter, then had notable presystemic extraction of the drugs, reducing bioavailability. Reptiles also have the renal portal system, which moves blood from the back half of a reptile through the kidneys before returning to the heart. This system has been linked to filtering of medications before systemic dispersion happens. With the renal portal system in reptiles it is generally more effective to give injections in the front half of the patient. Ethically, with the information at hand it is in our patient’s best interest to effectively manage pain, or at least possible discomfort in these species.

More and more published data in reptile analgesia is coming to light as researchers and the veterinary communities are becoming more aware of the similarities in pain perception of reptiles compared to mammals.

This is also an important development as many reptile species are used in the research setting. This is clearly evident in the research with NSAIDS in reptiles. Only one study thus far testing for analgesic efficacy has been published using ball pythons with doses of meloxicam, the most commonly used NSAID in reptiles. Serpents in other opioid-related studies have been shown to have a strikingly different anti-nociception affect than in chelonians and lizards, bringing to light concerns of dosing and efficacy in the other species. An example of this is commonly occurring thermal burns in snakes. It has been theorized that the evolution of snakes with their limblessness could have altered the spinal opioid expression of receptors, as well as function.

A pharmacokinetic study with green iguanas showed seemingly appropriate plasma concentrations of NSAIDs does not have a direct and predictable correlation to clinical efficacy. Another example involving opioids, the best-accepted drug for pain relief, used butorphanol. Butorphanol is a commonly prescribed kappa agonist and mu antagonist in reptiles for pain relief. However, it had no effect in corn snakes and worked well in bearded dragons. In contrast, morphine, a mu agonist worked well in the corn snake and not in the bearded dragon or turtles. Aquatic turtles tend to have more delta-opioid receptors than mu-opioid receptors in the brain, but determination of the receptors in the spinal cord is still unknown.

Authors in many of the more recent opioid studies note respiratory depression in their papers at higher doses, which, unfortunately, tend to be the most effective in anti-nociception. This should be a careful factor in dosing our patients. While most reptiles can tolerate a lower respiratory rate and even tolerate periods of anoxia, for excellent tissue healing and perfusion a bradypnea patient may hinder a speedy recovery.

Delivery of analgesics to reptiles comes in many of the same forms as with mammalian administration. Subcutaneous, intra-muscular, and oral administrations are the more common methods. Reptiles for oral administration of medications may need to have their mouth manually opened with a guitar pick or credit card. Although somewhat controversial, IM injections into the hind end of a reptile may be less effective due to the renal portal system. Intra-venous access is also possible in reptiles but is less practical and more difficult. Reptiles also can be given intra-coelomic injections.
Analgesia in Amphibians

Amphibians, unlike reptiles, go through different stages of metamorphosis during their life span. Amphibians go from having gills to lungs, being aquatic to semi-aquatic, having no legs to four or two and now with environmental contaminants sometimes six, more or less. This means at two different times in their lives we may treat amphibians differently. At one point like a fish and another more like any other terrestrial creature.

Amphibians are considered a “lesser vertebrate”, which means, as mentioned above; they fall lower on the evolutionary development chain. With this said, amphibians still do possess a well-developed endogenous opioid system with corresponding binding sites, primarily kappa-like receptors. Not only do opioids provide analgesia, but also, there has been potential shown with other drugs like ketamine, alpha 2's and tricaine methanesulfonate. It is important to note that amphibians have a reduced rate of hepatic metabolism of certain medications and appear to require greater doses of opioids for systemic effect than mammals. Although nociceptive receptors and the seemingly effective treatment of perceived pain exists with amphibians there is but one problem. There exists no thalamus to cortex networking to the cerebral cortex. This important anatomical neural feature makes is first appearance in reptiles. What this means is, amphibians still feel pain, but it may be much different than the way you or I may appreciate it.

One of the advances in veterinary pain management is the use of transdermal patches that deliver analgesia through the skin. In amphibians this has always been the case. Common anesthetic practices include anesthesia administration through the skin and thus is the same with analgesics. Common routes used in reptiles can also be used in amphibians.

Analgesia in Exotic Small Mammals

Pain management in these species is similar to that in the cat and dog with the exception of handling and restraint as well as dosing. Dosing ranges vary from text to text with ever changing research into efficacy of drugs and dosages thus far described. It is imperative for the reader to constantly be researching the newest most supported methods in pain management for whatever species one is working with. The recent publication of the mouse and rat facial pain-scoring chart is a crucial addition to any exotics practitioners’ library.

Sites for analgesia delivery vary from specie to specie and feasibility depending on muscle mass for IM injections, vascular access for IV injections and availability of SC space.

Rabbits are common pets and used extensively in the laboratory setting. In the laboratory setting, rabbits are often used as epidural and intrathecal models, and thusly this is a form of analgesia that can be utilized in these species. Signs of pain in these species mirror that of rodents, with the addition of hair plucking or ileus. Ileus can be intensified by the addition of opioids. Lagomorphs tend to be a sensitive species to chemicals of any sort, including analgesics. Fortunately more species-specific research is being performed finding more definitive dosing and relying less on extrapolation. One of the most recent studies published found previous meloxicam doses may be not as effective as once thought. The study highlights the importance of constant and relevant research into pharmacokinetics for all species.

Insectivore analgesia lacks a thorough research and practitioners are urged to investigate analgesia protocols for these species as more information becomes published. It is common practice to extrapolate doses of pain medications from rodent models. One published dose of buprenorphine is available at 0.01-0.03mg/kg SC q12hr for hedgehogs.

Much like insectivores very limited published data is available for marsupials and their 260 represented species in the world. In general marsupials tend to need higher doses of anesthetics and presumably analgesics for unknown reasons.

<table>
<thead>
<tr>
<th>Species</th>
<th>Vocalizing</th>
<th>Posture</th>
<th>Locomotion</th>
<th>Temperament</th>
</tr>
</thead>
</table>

**SPECIES-SPECIFIC BEHAVIORAL SIGNS OF PAIN**
<table>
<thead>
<tr>
<th>Animal</th>
<th>Squeak/Noise</th>
<th>Posture/Movement</th>
<th>Additional Symptoms</th>
<th>Behavior</th>
</tr>
</thead>
<tbody>
<tr>
<td>Mice, rats,</td>
<td>Squeaks, squeals</td>
<td>Dormouse posture; rounded back; head tilted; back rigid</td>
<td>Ataxia; running in circles</td>
<td>Docile or aggressive depending on severity of pain, eats neonates</td>
</tr>
<tr>
<td>hamsters</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Rabbits</td>
<td>Piercing squeal on acute pain</td>
<td>Hunched; faces back of cage</td>
<td>Inactive; drags hind legs</td>
<td>Apprehensive, dull, sometimes aggressive depending on severity of pain; eats neonates</td>
</tr>
<tr>
<td>Guinea Pig</td>
<td>Urgent repetitive squeals</td>
<td>Hunched</td>
<td>Drags hind legs</td>
<td>Docile, quiet, terrified, agitated</td>
</tr>
<tr>
<td>Chickens</td>
<td>Gasping</td>
<td>Stand on one foot, hunched huddled</td>
<td>None</td>
<td>Lethargic, allows handling</td>
</tr>
<tr>
<td>Birds</td>
<td>Chirping</td>
<td>Huddled, hunched</td>
<td>From excessive movement to tonic immobility depending on severity of pain</td>
<td>Inactive; drooping, miserable appearance</td>
</tr>
<tr>
<td>Fish</td>
<td>None</td>
<td>Clamped fins; pale color; hiding; anorexia</td>
<td>None unless forced; if a schooling fish; will separate itself from others</td>
<td>First sign to occur is anorexia; lethargic; stressed easily</td>
</tr>
<tr>
<td>Amphibians</td>
<td>None</td>
<td>Closed eyes; color changes; rapid respirations</td>
<td>Immobility; lameness</td>
<td>Anorexia; aggressive;</td>
</tr>
<tr>
<td>Reptiles</td>
<td>Hiss; grunting</td>
<td>Hunched; hiding; color change</td>
<td>Immobility unless forced</td>
<td>Anorexia; aggressive; lethargic; avoidance</td>
</tr>
</tbody>
</table>

This chart is meant to display some of the different signs species may exhibit if in pain. Individuals may not show any of these signs or they show signs not listed. This is meant as a general guide.


Fluid therapy is a fundamental practice in both human and veterinary medicine. However, in exotics a large discrepancy exists compared to our practices in small animal species such as dogs and cats. As exotic animals become more and more popular, owners will also depend on better treatment options for their unique pets, appropriate fluid therapy being one of them. Often by the time an emergent exotic species are brought to the veterinary clinic they are already suffering from moderate to severe dehydration and/or hypovolemic shock. These two conditions usually are not acute unless from severe blood loss secondary to blood loss (See Exotic Animal Blood Transfusion proceedings) but are chronic and have been hidden by the patient's own instinctual masking effects. In the small animal world, this would warrant aggressive fluid therapy, and theoretically does too in the exotic pet (exotic mammal, bird, reptile/amphibian). However, here lie a few problems. Starting with the lack of physiologic research. Little published sources of complete fluid studies are available. Rather, many guidelines are extrapolated from small animal sources and anecdotal evidence. Species diversity is another binding factor in well-documented fluid guidelines for exotic animals. There are thousand upon thousands of different species with ever so intricate physiological differences. Lastly, is the challenge of IV access. Intravenous access in stressed and decompensated small exotic animals comes with its own life-threatening risks from handling and the increased stress hormone release alone. Another fatal consequence is iatrogenic overhydration. This leaves practitioners having to weigh out the risks to benefits. Fluid therapy is indicated for any of the following reasons.
1. Replacement of fluid loss caused by dehydration, disease, hemorrhage, etc.
2. Correction of acid-base imbalances caused by various pathology and disease states
3. Correction of electrolyte disturbances caused by various diseases
4. Prevention of fluid loss during anesthesia, to maintain venous access, renal perfusion, blood pressure and counter act vasomotor effects of anesthetic drugs.
5. Maintenance of body fluid and electrolyte balance during on-going losses until the animal can compensate appropriately by water and caloric consumption
6. Administration of pharmaceuticals and parenteral nutrition.

Crystalloid and colloids fluids are two fluid types used in fluid therapy. Crystalloids are water-based solutions that come in a variety of solutions, some mimic electrolyte concentrations of plasma. There are three acting types of crystalloid fluids. "Isotonic" fluids have an equal osmolarity to surrounding erythrocytes and don't change fluid exchange across cell membranes. Isotonic crystalloids are effective at expanding the intracellular space. "Hypotonic" fluids actively increase erythrocyte volume and "Hypertonic" fluids pull water away form erythrocytes, decreasing their volume temporarily. Crystalloids are composed of smaller molecules and are osmotically active throughout body fluids. Crystalloids are capillary membrane permeable.

Colloids are also water-based but are a much larger molecule. They can be natural based or synthetic and usually stay within the vascular space. Colloids can only be given IV or IO. Compared to naturally derived colloids found in plasma, whole blood and albumin, synthetic colloids have an extended effect on intravascular volumes.

Synthetic colloids also offer an extended intravascular volume expansion effect and can reduce crystalloid total volumes by 40-60% when used in conjunction with each other. Their negative charge attracts water and sodium and are infused at much smaller volumes compared to crystalloids either by bolus or CRI. Most synthetic colloids do not provide plasma proteins or hemoglobin, however Oxyglobin™ and other synthetic blood products offer good volume expansion with the added benefit of tissue perfusion. Synthetic blood products have been used in birds and reptiles without complication. A negative attribute to Hetastarch™ is its effect of prolonging active clotting times.

As mentioned, hypovolemia is a common condition with exotic animals. Osmotic forces in the interstitium at >5% dehydration begin to pull water away from the intravascular space, which then effects perfusion. Filling the intravascular space with fluid that can be pulled into the interstitial space is key. It will also aid in cardiovascular support. Remember poor perfusion also means poor oxygenation! Normovolemia is essential to correct prior to hydration therapy.

Goal driven fluid therapy is important in all species, but even more so in the less understood exotic animal to avoid overhydration. Commonly, goal driven fluid therapy is defined as giving the least amount of fluid to achieve certain endpoints within normal vital sign ranges for a given species. Some endpoints include but are not limited to: Blood pressure, capillary refill time/ mucous membrane color, pulse quality, body temperature, mentation, heart rate, respiratory rate and urinary output.

*Blood component products and concentrated albumin are also effective means of intravascular expansion, but may not be practical in the exotic animal setting.

Fluid therapy in general is fairly imprecise and even more so with exotics. Every animal is constantly undergoing compartmental volume fluctuations combined with varying "ins and outs" based on the patient's willingness to consume water and/or disease processes. Three principles exist that are rather constant among all species of animals when fluid therapy is indicated. The first is fluid resuscitation as described above. This step may require a variety of fluid types (i.e. crystalloids and colloids). If cardiac conditions allow this initial step for chronic conditions can be done relatively safely in 12-24 hours with close monitoring. In more acute situations, the replacement stage can be done in as short as 1-4 hours, again with close monitoring for fluid overload. Step two is hydration. This entails replacing the interstitial fluid. Lastly, is maintenance and accounting for continued losses from a variety of modes (respiratory, salivary, urine output, blood loss, weeping wounds, etc.). Maintenance fluid rates are where many exotic animals usually start and ends, instead of the first two points in
goal driven fluid therapy. The maintenance rates are usually only ran during surgical procedures or a short time while the animal is in hospital. This stems largely from practitioner's unfamiliarity with a species and an unknown appropriate fluid therapy protocols. Sadly, this leaves many exotic pets still in a state dehydration and hypovolemia.

<table>
<thead>
<tr>
<th>NAME</th>
<th>Fluid</th>
<th>Dose IV or IO</th>
<th>Comments</th>
</tr>
</thead>
<tbody>
<tr>
<td>CRYSTALLOID</td>
<td>Replacement</td>
<td></td>
<td></td>
</tr>
<tr>
<td>0.9% Saline</td>
<td>Crystalloids</td>
<td>50ml/kg/day</td>
<td>Anesthesia rate: 5-10ml/kg</td>
</tr>
<tr>
<td>Lactated Ringer's</td>
<td>Colloids (Hetastarch, Oxyglobin)</td>
<td>10-15ml/kg</td>
<td></td>
</tr>
<tr>
<td>Plasmayte-A® pH 7.4</td>
<td></td>
<td>5ml/kg +15-40ml/kg with crystalloids</td>
<td></td>
</tr>
<tr>
<td>Normosol-R®</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>7.0% saline</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>5% Dextrose</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Maintenance</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>2.5% Dextrose in 1/2 strength lactated Ringer's</td>
<td>Crystalloids</td>
<td>10-30ml/kg/day</td>
<td>Over 72-96hr</td>
</tr>
<tr>
<td></td>
<td>Colloids (Hetastarch)</td>
<td>*15-25ml/kg/day intracoelomic &amp; epicoelomic</td>
<td>*Warm water baths are effective in partial rehydration with fluid absorption from the cloaca, it also may stimulate drinking</td>
</tr>
<tr>
<td></td>
<td></td>
<td>5ml/kg, not to exceed 40ml/kg/day</td>
<td></td>
</tr>
<tr>
<td>Natural</td>
<td>Crystalloids</td>
<td>100-150ml/kg/day</td>
<td>Repeat doses PRN</td>
</tr>
<tr>
<td>Whole blood</td>
<td>Colloids (Hetastarch)</td>
<td>5ml/kg</td>
<td></td>
</tr>
<tr>
<td>Frozen plasma</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Synthetic</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>8% Hetastarch</td>
<td>Crystalloids</td>
<td>60-120ml/kg/day (Small rodents 60-70)</td>
<td>The addition of colloid may be indicated</td>
</tr>
<tr>
<td>10% Panthastarch</td>
<td>Colloids (Hetastarch)</td>
<td>5ml/kg over 5-10 minutes</td>
<td></td>
</tr>
<tr>
<td>Dextran 70</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>Colloids (Hetastarch)</th>
<th>10-20ml/kg/day 5ml/kg for shock over 15 minutes</th>
<th>Do not exceed 20ml/kg/day</th>
</tr>
</thead>
<tbody>
<tr>
<td>Amphibians</td>
<td></td>
<td></td>
</tr>
<tr>
<td>1-2 parts balanced electrolyte crystalloid in one part 5% dextrose</td>
<td>25ml/kg intracoelomic</td>
<td>Use this method only in dire circumstances</td>
</tr>
</tbody>
</table>

**Baths for amphibians:**
- Amphibian Ringers Solution
  - NaCl 7.3g, KCl 0.17g, CaCl2 0.17g, NaHCO3 0.22g in 1 liter distilled water
- Whitaker-Wright Solution (100% solution)
  - NaCl 113g, MgSO4H2O 8.6g, CaCl2 4.2g, KCL 1.7g in 1 liter distilled water
- Hypertonic Saline
- Regular Saline (terrestrial species only)

**Important Formulas:**

**Correction of dehydration:**

\[ \% \text{ dehydration (estimate)} \times \text{Body weight (kg)} \]

**Total amount of fluid to give:**

maintenance + replacement (\% dehydration) + ongoing losses

**Calculation of osmotic pressure:**
2[Na] (meq/L) + glucose/18 (mg/dl) + BUN/2.8 (mg/dl) = plasma osmotic pressure.

Blood transfusions are an ever-growing therapy in the small animal practice. Transfusions offer patients a lifesaving option by giving us time to treat the underlying disease or correct hemodynamic imbalances. However, did you know you could use most of the same methods used in cats and dog blood transfusions in other species? Exotic animals suffer from anemia’s or require transfusions due to acute blood loss just like any other animal. However, transfusions are often overlooked in these species because of unfamiliarity and practicality.

Whenever transfusing a patient many constants exist. Donor selection is very important. Selecting a healthy donor from the same species is ideal. Even better, is if the donor is from the same home to reduce the risk of cross infection for a number of diseases. A stressed donor with a release of cortisol can be avoided with light to moderate sedation. It is recommended that if sedation is used (in most cases will be) in the donor animal, all the anesthetics/sedatives should be listed and chosen with the recipient’s health status in mind. Residual effects have been described in recipient animals from medications given to the donor. Blood substitutes are still available outside of the USA and have been used in all of the species described in these notes. These products (Oxyglobin™, Hemopure™, Dextran-Hemoglobin™) may offer an advantage to the practitioner by allowing a constant supply and not having to rely on the time for collection, stress, risk factors and accessibility of a donor animal.

Monitoring and transfusion rate guidelines during a transfusion do not change among species. Although slight variances occur, many guidelines have all vital signs (including mentation) being recorded every 5-15 minutes for the first 30 minutes. The starting transfusion rate is 0.25ml/kg for that first 30 minutes and can be increased to a rate that would make the whole transfusion completed within 4 hours. After the first 30 minutes of the transfusion, with no reaction, the monitoring frequency can be decreased to every 15-30 minutes. Remember, guidelines are merely guidelines! Patients may need more or less attention and by the means of slower or quicker transfusion rate.

In many of the species discussed below, in particular the non-mammals, blood compatibility is less of a concern especially if this is the patient's first transfusion. The lessened concern stems from the lack of identified blood groups in bird and reptilian species. It is encouraged to perform a major and minor crossmatch.

**Reptilian and Amphibian Blood Transfusion**

Reptiles and amphibians are one of the least common species one will have to perform a blood transfusion on. These species have amazing regenerative attributes and can tolerate some pretty inhospitable environments. These attributes also hold true for their survivability of medical conditions. Few studies and just as few clinical write-ups have been done on transfusions in these species. Most of the literature leans away from transfusing an amphibian all together, but rather treat the underlying condition and hope for the best.

Reptilian blood transfusions, on the other hand, are a more feasible therapy. A general guideline for most species suggests transfusions when the PVC drops below 20%. This is a good guideline, but many nucleated species can tolerate lower PCV's, and a careful clinical evaluation should be made before jumping to a blood transfusion.

The morphology of reptilian and amphibian red blood cells include a nucleus in an oval shaped cell. The erythrocyte size is also one of the largest in the animal kingdom, which explains a lower PCV compared to mammals.

*Brumation can have associated low PCV and is a normal finding*

Setup and precautions are the same as with mammal transfusions, but IV access poses a bigger challenge. In many reptiles, cut downs are often necessary for basic indwelling IV catheters. Common sites for IV catheterization include the jugular for chelonians (which can be done percutaneously), tail
vein, and in large lizards cephalic or saphenous veins. In more critical and non-ambulatory patients the ventral abdominal vein is a useful site.

Collection sites from a donor include all of the before mentioned, but also include the heart in certain reptiles such as snakes. An often more practical means of administration and easier accessibility in reptiles is by using an intraosseous catheter. In lizards, the preferred site is the tibial crest. Sites such as the proximal humerus, femur and ulna can also be used. Snakes obviously are not candidates for IO catheters. Reptile and bird blood can clot quickly and using a heparinized syringe with sodium citrate may be indicated instead of using CPD, CPDA or ACD. *Heparin can cause RBC lysis in chelonian blood. Although, not ideal cross-species transfusions have been described in the literature. More rapid cell death often occurs, and the potential for cross-reactions is greater. Because normal reptilian erythrocytes have a long life span, 600-800 days, a quicker die off of heterologous transfusions may need to be repeated, increasing a patient's chance for reaction, stress and infection. When transfusing ANYTHING, a blood filter for clots is vital! One study on alligator blood determined that the 18-micron filter did not lead to hemolysis or a decrease in PCV with smaller quantities of blood, suggesting the efficacy of the filter in other reptilian species.

*polychromasia is not uncommon in shedding reptiles
*EDTA can cause hemolysis in some reptile and bird species. Lithium heparin is recommended instead for hemograms

**Avian**

Avian blood transfusion medicine is nearly the same as reptilian. The morphology of the avian erythrocyte is almost identical to the reptile RBC but is a bit smaller. The transfusion guideline of a PCV below 20% also applies to avian species. Birds are more tolerant of lower PCV's especially if chronically acquired. One author has even described seeing patients with PCV's ranging from 7-12%! They also describe not transfusing the patient if the bird is bright, alert and reactive. In studies with ducks, the investigator found hypovolemic shock only occurred after 60% acute blood loss. In another study, 50% loss of total blood volume was corrected without blood product resuscitation, but instead with crystalloid and colloid fluids. As with any species, homologous transfusions are preferred, but pigeon to raptor transfusions have been successful, although erythrocyte life was short. Erythrocyte life span is only 28-45 days in birds, couple this with a small spleen that does not act as an RBC reservoir, and avian patients can become anemic again quickly. Unlike the reptile, erythropoiesis can occur rapidly, and reticulocytes can occur within hours. With varying degrees of anemia more pronounced, RBC size variances might occur; however, cell size has a normal "cell distribution width" of 10-11%. Even with an increased erythrocyte size compared to mammal's one study in chickens concluded the 18-micron filter does not cause significant hemolysis.

*Poikilocytosis (Variable cell shape) is most commonly seen with systemic disease that affects bone marrow or is an artifact error.

*A bird in respiratory distress with an anemia has a greater need for a blood transfusion. Birds have a more efficient respiratory system, therefore, decreasing the amount of RBC's needed for carrying oxygenated hemoglobin. If this mechanism is altered, greater demand for RBC's is warranted.

Blood groups among birds have not been thoroughly studied among many species. However, in chickens twenty-eight different blood group antigens have been described. Multiple other blood groups have discovered in other common species such as turkeys, pheasants, quail and ducks. Sedation or light anesthesia is more than likely going to be needed for blood collection from a donor and is often the
safest and least stressful approach. As with any species choosing sedatives that will have minimal effects on the donor and the recipient is ideal. Collection and transfusion sites in the bird include the right jugular (careful attention should be paid so that the adjacent air sacs are not cannulated), ulnar vein, brachial vein and the medial metatarsal vein. The recommended volume is 1-2% of the patient's body weight. Sodium citrates is the recommended anticoagulant in birds; however ACD, CPDA and CPD are also compatible. Heparin has also been described at 0.25ml (1000U/ml Heparin) to 10ml of whole blood. Intraosseous catheters maybe indicated in severely hypovolemic patients. The humerus and femur should be avoided for any IO catheter in the avian patient as they are pneumatic, and one can drown the patient if fluids are given through these sites. Commonly IO catheters are placed in the tibiotarsus or distal ulna under appropriate anesthesia/ analgesia.

Other Indications for transfusions:

Coagulopathies in reptiles and birds do happen. However, treatment is limited to treating the underlying inflammatory response. Accurately estimating clotting times in reptile and avian species is still being studied. The literature has described methods such as PT and PTT as possible reliable clotting indicator tests. Methods such as ACT and whole blood clotting times are more reliable at this time. Blood component transfusions in theory are possible, but can prove impractical in most settings.

Protein imbalance disorders are also not uncommon disorders. Reliable tests for determining true serum or plasma protein levels are still evolving. Thankfully present research has shown refractometry is fairly accurate and is a device most practices already have. It should be noted plasma protein will be higher in bird and reptile species due to the presence of circulating fibrinogen. Hyperproteinemia can be associated to egg production in both birds and reptiles. Other etiologies for hyper and hypoproteinemia seen in mammals are also causative factors in both special species. Total protein levels in the reptile range from 3-7 g/dL and 3-4g/dL in birds. Birds of prey usually have a slightly higher plasma protein compared to their non-carnivorous cousins.

Exotic Small Mammals

There are relatively few differences between methods used in cat and dog transfusion medicine to exotic mammal transfusion medicine. The biggest differences exist with the overall accessibility of donor animals and feasibility of the transfusion in practice. Acute blood loss of 30-40% total blood volume will lead to shock and fluid resuscitation is warranted. When more than 40% total blood loss occurs immediate volume resuscitation is warranted. If the corrective actions do not lead to appropriate tissue oxygenation an immediate blood transfusion or blood substitutes need to be considered. A benefit of blood substitutes like Oxyglobin™ includes a smaller molecule size giving it the ability to better access micro capillaries for improved perfusion. It also has a 10 fold better resuscitative effect in hypovolemic shock than actual blood because of vasoconstricting properties, therefore, requiring less volume to be infused.

The average RBC lifespan of a small exotic mammal is relatively short (22-55 days) in comparison to a dog (100-120 days).

*Howell-Jolly bodies are normal in the rabbit and do not indicate cellular regeneration as seen in the feline patient

Sedation is also a common practice is the exotic mammal to reduce stress, injury and hormone release. Intravenous and intraosseous catheterization is well described for many species of small exotic mammals.
Micro hemofilters used in small animal practice are effective and recommended in the exotic small mammal. Little research has been done into defining different blood groups in many small exotic mammal species. A major and minor crossmatch should be performed before any transfusion with special attention to the more important major cross match.

*It should be noted that volatile anesthetic gasses, such as isoflurane and sevoflurane, have been associated to a significant drop in hemoglobin and hematocrit levels in ferrets.

References available upon request