



UNIVERSIDAD NACIONAL MAYOR DE SAN MARCOS  
FACULTAD DE MEDICINA VETERINARIA

Lima, Perú



# The Latin American Veterinary Conference TLAVC 2006



*Programa de Educación  
Continua para América  
Latina del North American  
Veterinary Conference*



*29 y 30 de Setiembre  
1 y 2 de Octubre*



# RESUMEN

animales de compañía



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## ANESTHESIA IN EXOTIC PETS

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There are books written on the subject of anesthesia in all the different species. However, in private clinical practice, it is not necessary to know fifty ways to anesthetize a hamster. What is needed is a standard protocol for the average patient. A standardized protocol should be established for all the different patients that are treated by your hospital. Whether the anesthesia is performed by a veterinary technician or by the veterinarian, it is important that these protocols are followed. Once you get used to a particular drug or anesthetic protocol, you can learn what to expect, and will be better able to deal with problems should they arise.

In general you should not vary from these routines unless a specific situation dictates the need. The following protocols are those that the authors have used with great success in exotic practice.

### GENERAL CONSIDERATIONS

#### Fasting

Because of their small size and rapid metabolisms, it is generally not recommended to submit small mammals to a prolonged fast prior to surgery. To minimize the risk of regurgitation and aspiration, it may be wise to withhold food for no more than three hours prior to the procedure. Guinea pigs will store food in their mouths, and it is not uncommon for them to spit this food up after they are induced. Careful swabbing of their oral cavities with cotton swabs will usually suffice.

Birds have relatively little stores of glycogen in their livers, and as a result, will experience rapid hypoglycemia if fasted for prolonged periods. The three hour limit is appropriate for avian patients as well.

Reptiles do not have a problem with regurgitation during anesthesia. Therefore, it is usually not necessary to fast these animals prior to surgery unless you are planning on a gastric procedure, in which case a fast and an enema may be essential.

#### Pre-operative

All of the general principles of anesthesia and pre-operative evaluation that apply to mammal patients also apply to exotic pets. A thorough pre-operative physical examination by the surgeon, appropriate laboratory screening, radiographs etc., should all be performed, or at least recommended to the client. Patients should always be classified as to their risk of anesthesia (healthy pet, elective procedure vs. patient with major health problems), and the potential surgical risk (simple restraint for laboratory sample collection vs. prolonged procedure with potential for major blood loss).

Caution should be taken with patients having a low PCV, as these animals do not have a lot of blood to start with, so even minor hemorrhage can be lethal in a small pet. Consider pre-operative transfusions in those animals at risk, have blood ready for intra-operative transfusions, or at least pre-medicate patients with fluids prior to the procedures.

Since hypothermia is of paramount concern in these little patients, the fluids should always be pre-warmed prior to administration. In addition, due to the generally higher metabolic rates in these little animals, it is wise to use a 5% dextrose solution IV, or a 2.5% dextrose in a balanced electrolyte solution either pre-operatively, intra-operatively, or immediately post-operatively.

Most of the exotic pets that we deal with have a relatively large surface area to volume ratio (in general, the smaller the pet, the larger the ratio). As mentioned, this larger surface area means that these patients are prone to hypothermia. Some form of supplemental heating is mandatory for these patients. Circulating warm water blankets, water filled gloves (USE EXTREME CAUTION WITH THIS

TECHNIQUE!), warmed anesthetic gasses (tube warmers), heated fluids, heated lavage fluids, and for surgical prep, the use of chlorhexidine rather than alcohol.

The avian patients, with their unique respiratory system, are also prone to hypothermia. Since, in order for a bird to respire, gas needs to constantly flow across the parabronchi, there is considerable potential for heat loss. Again, many of the above principles for preventing hypothermia apply.

Reptiles, being ectothermic, can be totally unpredictable in their response to anesthesia. To better control this, it is best to eliminate temperature as a variable. Every reptile group has its own preferred optimal temperature. For instance, a garter snake thrives at temperatures around the high 70's to the low 80's, whereas the green iguana needs temperatures in the low 90's to flourish. So, when performing anesthesia in these patients, where a garter snake may do just fine at 80 degrees, the iguana may be slower to induce, may be more difficult to maintain, and may take much longer to recover.

## **Induction and Anesthesia**

### ***Small mammals***

Injectable anesthetics are routinely used in small mammal anesthesia. These will often provide good restraint and anesthesia for short procedures. However, keep in mind that some of the injectables, such as ketamine, provide questionable analgesia.

Combination drug use (called cocktail anesthesia) is preferable to single drug administration. Every drug has its advantages and disadvantages. Using combinations tend to take advantage of the best qualities of each.

Ketamine, a cyclohexamine dissociative (cataleptic) anesthetic, is one of the most commonly used injectable anesthetics in veterinary medicine. It anesthetizes through STIMULATION. It is highly lipid soluble, allowing it to quickly enter the brain tissue. It induces paralysis and muscle rigidity.

Reflexes, such as swallowing, coughing, pedal and corneal, are maintained. However, animals under ketamine lose their blink or palpebral reflex. As a result, the cornea needs to be lubricated to prevent drying out.

The big disadvantage of ketamine is the risk of convulsions during recovery. To ameliorate this, combination anesthesia with a tranquilizer, such as diazepam (valium), xylazine (rompum) or acepromazine (ace) is recommended. These can be administered either as a pre-medication, or mixed and given at the same time.

Animals under ketamine influence will salivate. Administering an anticholinergic, such as atropine or glycopyrrolate, will help with this side-effect. Glycopyrrolate is more effective at controlling secretions than atropine, and is less likely to cause tachycardia or cardiac arrhythmias. In addition, many rabbits have an «atropinesterase» that renders atropine useless.

For prolonged (more than 5 minutes) procedures, or potentially painful procedures, the patients should be maintained on a volatile anesthetic such as isoflurane.

Ferrets are readily intubated, and for any procedure that is involved or prolonged, this should be done for safety. Although all rodents are capable of being intubated, doing so can be a challenge in most species.

Guinea pigs have a palatal ostium that blocks easy access to the glottis. This can be gently diverted, and should be done for difficult or prolonged procedures. In addition, guinea pigs have a tendency to store food in their cheeks, and care should be taken to swab this out prior to anesthesia to prevent potential aspiration.

Rabbits are perhaps the most difficult to intubate. We do not typically intubate rabbits for short procedures, such as neuters, but always intubate for any prolonged or difficult surgery.

There are talented individuals that are capable of intubating rabbits blindly, by passing the endotracheal tube either down the pharynx or through the nostril, and pass it directly into the glottis by «listening» to the

breath sounds. By timing the passing of the tube with inspiration, they are able to slip the tube into the glottis.

For those less gifted, using a neonatal laryngoscope or, an otoscope with a large dog speculum, you can readily visualize the glottis while the animal is under anesthesia. Then, a polyethylene urinary catheter is placed through the otoscope cone and into the open glottis. The cone is then removed, leaving the stylet behind. The endotracheal tube is then threaded over the stylet and into the trachea. The stylet is then removed.

An alternative to general anesthesia in rabbits is epidural anesthesia. It has many benefits, and is not difficult to perform.

Epidural anesthesia and epidural analgesia are techniques whereby medications are injected into the epidural space, the potential space around the spinal cord inside of the vertebral column. Analgesia is the injection of local anesthetics. Anesthesia is the injection of medication such as opiate agonists and alpha 2 agonists into the epidural space. Spinal analgesia is the injection of a local anesthetic into the subarachnoid space as it mixes with the CSF.

There are a number of advantages of epidural anesthesia. Epidural administered anesthetics and analgesics are used for the management of surgical cases, obstetrical pain, and post surgical pain and chronic pain. Epidural drugs administer pain relief with less drug compared to systemic administration. This is important when the administered drug has negative side effects such as cardiac and respiratory inhibition. We have seen epidural medications decrease recovery time. We also have used it in place of systematically given post surgery pain relief. It not only seems to be more effective than systematically administered drugs, but there are less negative side effects, including sedation. It is important to have a surgical protocol that allows for pregnant guinea pigs to be minimally stressed so they don't become anorectic, develop post surgical ketosis, and minimizes the depressive effects of medications on the dam and young. Epidural medications accomplish this better than systematically injected pain relievers. In rabbits, there was no change in heart rate with epidural lidocaine.

There are some disadvantages with epidural medication. For most of our small mammals, an epidural catheter cannot be placed. Only a needle can be inserted into the intervertebral space. This is because of the small size of epidural and intervertebral spaces and the cranially directed dorsal spinous process overlying the intervertebral space. If a catheter is not placed, it is difficult to administer continuous epidural medication. If the epidural block extends into the cervical region, it may block phrenic nerves and cause respiratory arrest. Incorrect technique can cause trauma to the spinal cord. Inadvertent subarachnoid deposition can cause greater cranial advancement of the drug.

Local anesthetics are the most commonly used drugs for epidural analgesia but such drugs as opiate agonists, alpha 2 agonists, and ketamine have also been used. Either a catheter or a needle is used. We have only used a needle and there has not been a need for a epidural catheter placement. The cranial extent of the epidural analgesia depends the extent of intervertebral foramina occlusion. Occlusion is caused by calcification, fat, and fibrous tissue and the more occlusion, the further cranially the analgesia is present. If the drug is injected into the large venous plexous on the floor of the vertebral canal, higher than expected levels will enter the blood stream. After epidural injection of lidocaine in most species, analgesia develops within 5 to 15 minutes and lasts from 60 to 90 minutes. Bupivacaine can provide up to 4.5 to 6 hours of surgical analgesia. Some published doses include: guinea pigs with 1% lidocaine at 0.25 ml or 2% lidocaine at 0.3 ml. Guinea pigs have been given bupivacaine 0.25% at 0.12ml/kg. And rabbits have been treated with lidocaine 1.5% at 0.4ml/kg. It is best to use drugs without preservatives so as to minimize adverse reactions.

Our protocol begins after the patient is anesthetized for the procedure. We have used epidural anesthesia commonly for abdominal procedures and for one thoracic procedure. The intervertebral space is located and the area is surgically prepared. A 25 gauge needle is inserted through the skin and is further inserted. Cerebrospinal fluid is seen when the needle is in the subarachnoid space. When the needle is in the epidural space, there is no fluid and a drop of flush easily enters the space. The medication is slowly injected and then it is followed by a small amount of flush.

## **Birds**

Injectable anesthetics tend to be unpredictable, and have been used with limited success. Ketamine, ketamine combined with valium or ketamine combined with xylazine, are the most frequent drugs used.

These provide restraint and light anesthesia for short, minimally invasive or painful procedures, but, the patients tend to experience poor muscle relaxation, wing flapping and prolonged recoveries. With rare exception, isoflurane is the restraint/anesthetic method of choice in birds, even for short procedures.

Pre-medications such as tranquilizers or anticholinergics are rarely used. With the possible exception of extremely flighty birds, and some wild birds, pre-medications are not necessary.

Isoflurane is the volatile anesthetic of choice in birds. There is no justifiable reason to use the older gasses such as methoxyflurane or halothane. With halothane, the heart stops at the same time breathing stops, meaning that there is little to no margin of safety. Using isoflurane, the heart continues to beat following apnea for a short time, allowing an attentive anesthetist to take appropriate action.

Birds are readily masked down with isoflurane. Once unconscious, the mask is removed and the patient is intubated being careful to not damage the delicate trachea. In birds, the trachea consists of complete tracheal rings, so, the trachea will not distend when the cuff is inflated. As a result, I prefer to use shouldered cole tubes rather than cuffed tubes. If you use a cuffed tube, then select a tube that has a snug fit, and do not inflate the cuff.

The air sac system in birds provides a unique adaptation for anesthesia that is not seen in other species. Because of the one-way air flow pattern in bird lungs, a breathing tube can be placed through the bird's side into the caudal thoracic air sac. Using an oxygen flow of 1 l/min, the isoflurane is passed directly into the lungs and out the glottis. Since the gas is under constant motion, the bird will not actually breath during the anesthesia. This technique is useful for surgery of the head, neck, trachea and syrinx.

## Reptiles

Injectable anesthetics are used more frequently in reptiles than in birds. However, the agents are used more for restraint than for anesthesia.

Ketamine and Telazol (tiletamine/zolazepam) are the drugs of choice. Telazol needs to be reconstituted, and if not used within 14 days, goes bad. It has the advantage over ketamine in that the dose is much smaller, being especially useful in larger patients (large constrictors, tortoises).

Ketamine, due to its acidic pH, has a tendency to cause discoloration to the scales in reptiles when injected superficially. These marks usually result in permanent discoloration, so, it is prudent to advise your client prior to using it.

Reports from England suggest that Propofol makes an excellent pre-medicant in iguanids. At a dose of 1 ml/kg, IV in the tail vein provides 10 - 15 minutes of anesthesia, allowing for intubation or minor procedures. Keep in mind that Propofol does not produce analgesia. Recently, Bennet et. al recommends a dose of 5-10 mg/kg IO as an induction agent. The animals were then maintained at 0.5 mg/kg/min. Supplemental oxygen via an endotracheal tube was recommended.

As with the other exotics, for prolonged procedures, it is wise to maintain the reptile patient with a volatile anesthetic. Again, isoflurane is the agent of choice as it permits rapid induction and compared to the other agents, a more rapid recovery.

In most lizards mask induction is possible. Snakes and turtles can hold their breath for minutes to hours. If injectable anesthetic induction is not feasible, then placing these animals in an induction chamber, under supervision, is possible.

Reptiles are generally easy to intubate. In snakes and varanid lizards, the glottis is immediately inside of the mouth, making intubation an easy task, even in awake animals. In most lizards and the turtles, the glottis is caudal to the fleshy tongue. In these animals it is usually necessary to pre-medicant with Telazol or ketamine prior to intubation.

In reptiles, unlike in mammals, the glottis is always closed except during inspiration. This means that the anesthetist must be patient when performing the intubation. Do not force the tube into a closed glottis, rather wait until the patient takes a breath and then gently pass the tube into the trachea.

Snakes and lizards have a «C» shaped tracheal cartilage, the dorsal surface being comprised of a membrane. Turtles and the crocodylia have complete tracheal rings. When intubating snakes and lizards,

extreme care must be taken not to damage this delicate dorsal tracheal membrane. It is preferable to use shouldered «Cole» tubes rather than endotracheal tubes with a cuff.

Reptiles lack a diaphragm. Their breathing is voluntary, not involuntary as it is in mammals. This means that when they are fully anesthetized, they become apneic - or will stop breathing. They must be ventilated in order to maintain steady state anesthesia.

Induction is accomplished by administering one breath every ten seconds. The pressure should not exceed 15 cm H<sub>2</sub>O (11 mm Hg). Once induced, maintenance can be accomplished with one breath every 30 seconds. Recovery can be achieved with no more than one breath every minute until spontaneous respiration returns.

The stimulus for reptiles to breathe is a decreasing oxygen tension, as opposed to mammals, whose stimulus to breathe is an increase in carbon dioxide. This is important for post-anesthetic recovery since it is imperative to discontinue the oxygen as soon as the procedure is finished. For recovery, continue to ventilate using only room air.

### **Post-operative**

The most important thing to remember post-operatively is to maintain the patient's warmth. The pet should be moved to a warmed recovery cage or incubator. In addition, fluid balance must be addressed, as dehydrated patients will have a more difficult time metabolizing or eliminating the anesthetic.

Post-operative analgesia should always be a consideration. Few analgesia studies have been done in birds and reptiles, but substantial work has been done in small mammals due to their extensive use in laboratory research. Several analgesics are listed at the end of the proceedings.

### **PATIENT MONITORING**

Over the years several monitoring devices have been employed to evaluate anesthetized patients. The electrocardiogram has been the gold standard. Although the significance of the tracings may not be fully understood for all the species, it usually provides reliable indication of the patient's pulse rate. In addition, to the observant anesthetist, it may also provide clues toward impending changes in the patient's condition. Many of the smaller patients, and most of the reptilian patients, are impossible to monitor with an E.C.G. due to either their rapid heart rates, or the small electrical potential produced.

Ultrasonic Dopplers detect pulsatile blood flow in the patient. It produces an audible signal for each pulse wave. It is considered very accurate, but has the disadvantage of not being able to provide any clues on the patient's physiological changes. In addition, the noise that the Doppler produces is grating to listen to, and is usually best when attached to headphones.

The stethoscope, regular bell type or esophageal, is an excellent monitoring device providing that it is used. However, they are usually ineffective in reptiles.

Recent advances in anesthetic monitoring in human medicine have seen the introduction of the pulse oximeter to veterinary anesthesia. Specifically, the pulse oximeter has been used to monitor pulse and oxygenation during anesthesia and critical care settings. The pulse oximeter employs non-invasive technology and is user-friendly, requiring minimal training for proper and accurate usage by even lay staff. These attributes make its use in anesthetic monitoring for exotic patients an attractive adjunct to the more conventional electrocardiogram or Doppler ultrasonic flow detectors.

The pulse oximeter is a spectrophotoelectric device that is applied typically via means of a clamp, preferable over a pulsating vascular bed to glabrous areas of the skin. The probe contains a diode that emits light in both the red and infra-red wavelengths. This dual-wavelength emitted light then passes through the pulsating vascular bed adjacent to the diode, and then is registered in a photodetector. The percent transmittance of the light is calculated within the unit and further equated to an oxygen saturation estimate.

Reptiles, unlike mammals which maintain respiratory drive based on many factors, including blood CO<sub>2</sub> and pH, regulate their respiratory rate by careful balance of oxygen partial pressure (PO<sub>2</sub>) and body

temperature. At higher temperatures, the tissue demand for oxygen likewise increases. The increase demand is not met by an increase in respiratory rate, but rather an increase in tidal volume.

Of particular clinical significance here is the ability to monitor the delivered oxygen to the tissues of anesthetized reptiles. It is not uncommon for reptiles post-procedure to remain anesthetized for prolonged periods. By monitoring SpO<sub>2</sub> it may be possible to more accurately assess the state of the reptilian patient. In mammalian anesthesia the tendency is there to frequently bag the patient during recovery, whereas, in the reptilian patient, this increased oxygen may actually be inhibiting the return to spontaneous respiration.

The sensor traditionally has been incorporated into a finger or «clothespin-like» clamp positioned either over the patient's finger (human) or tongue (veterinary patient). There are several new probes available for use with exotics (Surgivet). Aside from the standard tongue clips, there are now C-clamps, tail wraps and esophageal/rectal probes. Of all the species, the birds are probably the most difficult to monitor, however, it is usually possible, using one of the probes, to monitor most any patient. The esophageal probes seem to be the most effective in the avian patient.

Reptilian patients have been difficult to monitor due to their thick skin and oftentimes heavy pigment, making placement of any kind of clamp-on type of sensor ineffective. The rectal or esophageal probe make the measurement of pulse rate and arterial oxygen saturation more feasible even in these patients.

Placement of the esophageal probe is best when the spectrophotometric diodes are positioned adjacent to the internal carotids/jugular vein complex, which is readily accessible through the oral cavity in an anesthetized patient. In patients where this is not logistically feasible (oral surgery), a membranous sheath can be applied over the probe so that it can be inserted rectally, with the diodes directed dorsally so that it can sense the caudal aorta and renal arteries. This is not as efficient in the avian patients. In these cases, using the C-clamp over the thigh has been effective.

## **CONCLUSION**

Regardless of the instrumentation employed, there is no substitute for an alert, attentive anesthetist. Heart rate, respiratory rate, mucous membrane color, capillary refill time and reflexes are all reliable, useful parameters for monitoring the anesthetized patient. Don't forget your medicine that you learned in dogs and cats. Don't be afraid to apply the principles to your exotic patients.

## ANESTHESIA OF THE RABBIT

### Pre-operative

- PE
- Lab work (blood, radiographs, E.C.G., etc)
- IV, Fluids prn
- Antibiotics prn
- Pre- meds (glycopyrrolate, 0.01-0.02 mg/kg SC, IM)

### Induction

- Ketamine = Alone, not recommended
- or - Ketamine + xylazine = Mix equal volumes of ketamine (100 mg/ml) with xylazine (20 mg/ml).  
Administer 0.5cc/kg, IM
- or - Ketamine + diazepam = 20-40 mg/kg ketamine + 1-5 mg/kg diazepam, IM 10-20 mg/kg ketamine + 0.5 mg/kg diazepam, IV
- or - Ketamine + acepromazine = mix 10 parts ketamine (100 mg/ml) with ace (10 mg/ml). Administer 0.25 cc/kg, IM.

### Maintenance

- Epidural with lidocaine or oxymorphone
- Isoflurane via mask or endotracheal tube

### Anesthetic monitoring

- Stethoscope
- Monitoring reflexes
- ECG
- Doppler
- Pulse Oximeter

### Recovery

- Keep warm and quiet

### Analgesia

- Butorphenol, 0.1 - 0.5 mg/kg SC, IM, IV, q 2-4 hrs
- or - Flunixin meglumine, 1 - 2 mg/kg, SC, IM, q 12-24 hrs
- or - Epidural with lidocaine, oxymorphone

## ANESTHESIA OF THE RODENT

### Pre-operative

PE  
 Lab work (blood, radiographs, E.C.G., etc)  
 IV, Fluids prn  
 Antibiotics prn  
 Pre- meds (glycopyrrolate, 0.01-0.02 mg/kg SC, IM)  
 Empty/clean cheek pouches (hamster/guinea pig)

### Induction

Ketamine = Alone, not recommended  
 - or- hamsters/mice      rats      g. pigs      gerbils      chinchillas  
 Ketamine 50-200 mg/Kg, IM, IP 50-100 20-40 50-70 — + xylazine 5-10 mg/kg, IM, IP 5 5 2-3 —  
 - or - Telazol 50 -80 mg/kg, IM, IP 50-80 20-40 50-80 20-40  
 - or - Ketamine 50 - 100 mg/kg, IM, IP 50-100 20-50 50-100 20-50  
 + ace 2.5-5, mg/kg, IM, IP 2.5-5 0.5 2.5-5 0.5

### Maintenance

Isoflurane via mask or endotracheal tube

### Anesthetic monitoring

Stethoscope  
 Monitoring reflexes  
 ECG  
 Doppler  
 Pulse Oximeter

### Recovery

Keep warm and quiet

### Analgesia

Butorphenol, 0.1 - 0.5 mg/kg SC, IM, IV, q 2-4 hrs  
 - or - Flunixin meglumine, 1 - 2 mg/kg, SC, IM, q 12-24 hrs

## ANESTHESIA OF THE FERRET

### Pre-operative

PE

Lab work (blood, radiographs, E.C.G., etc)

IV, Fluids prn

Antibiotics prn

Pre- meds (glycopyrrolate, 0.01-0.02 mg/kg SC, IM)

### Induction

Ketamine = Alone, not recommended

- or -

Ketamine + xylazine = ketamine, 10-30 mg/kg, IM  
xylazine, 1-2 mg/kg, IM

- or -

Ketamine + diazepam = ketamine 10-30 mg/kg, IM  
diazepam, 1-2 mg/kg, IM

- or -

Telazol = 22 mg/kg

- or -

Ketamine + acepromazine = mix 10 parts ketamine (100 mg/ml) with ace (10 mg/ml). Administer 0.25 cc/kg, IM

### Maintenance

Isoflurane via mask or endotracheal tube

### Anesthetic monitoring

Stethoscope

Monitoring reflexes

ECG

Doppler

Pulse Oximeter

### Recovery

Keep warm and quiet

### Analgesia

Butorphenol, 0.1 - 0.5 mg/kg SC, IM, IV, q 2-4 hrs

- or -

Flunixin meglumine, 1 - 2 mg/kg, SC, IM, q 12-24 hrs

## ANESTHESIA OF THE REPTILE

### Pre-operative

PE

Lab work (blood, radiographs, E.C.G., etc)

IV, Fluids prn

Antibiotics prn

Pre- meds (glycopyrrolate, 0.01-0.02 mg/kg SC, IM)

### Induction

Ketamine = 50 mg/kg, IM

- or -

Telazol = snakes, lizards, water turtles, 5 mg/kg, IM

tortoises, 10 - 20 mg/kg, IM

- or- face mask induction = lizards

a personal computer, a modem, and a telephone jack (or broadband access) near the computer. Both IBM compatible and Macintosh computers may be used. Computer store dealers should be consulted for advice on brands, styles, and features.

The Veterinary Information Network (VIN) is available on the WEB with basic and comprehensive services offered at different subscription rates. For information go to [www.VIN.com](http://www.VIN.com). VIN also allows dialogue between subscribers and moderators. VIN includes exotic animal bulletin boards on reptiles, birds, and mammals other than dogs and cats. There is a fee for use when accessing VIN. Fees vary depending on the level and amount of use but start around \$50 US per month.

International Veterinary Information Service, or IVIS, is a FREE service that has a very large, international subscription. There is a tremendous amount of information available on all species. From the IVIS site you can search many different databases and download entire articles and proceedings from conferences. There is a fee for some of these advanced services. Go to [www.IVIS.org](http://www.IVIS.org) to register.

Of course there are many, many websites available for all the different species. It is important to be careful when collecting information from the web if you don't know the validity of the source.

### GENERAL

Merck Veterinary Manual. 2006; Merck & Co., Inc. Whitehouse Station NJ, USA.

There are several excellent chapters on exotics pets.

Manual of Small Animal Practice. Birchard and Sherding. Elsevier. St. Louis, MO. 2006.

There are several chapters on exotic animal pets. This is an excellent reference for the practice on a limited budget that needs a resource to cover all small animal topics.

\* The Veterinary Clinics of North America – Exotic Animal Practice. Published 4 times per year by Elsevier. St. Louis, MO.

\* Manual of Exotic Pets. Meredith A, Redrobe S. 4<sup>th</sup> edition. 2001. British Small Animal Veterinary Association. Worthing, West Sussex, 1991.

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\*Atlas of Diagnostic Radiology of Exotic Pets, Small Mammals, Birds, Reptiles and Amphibians. Rubel, GA, Isenbugel, E, Wolvekamp, P. W B Saunders Co., Philadelphia. 1993.

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**REPTILES**

\* Mader DR. Reptile Medicine and Surgery, 2<sup>nd</sup> ed. Elsevier Inc., St. Louis, MO. 2006. This is a comprehensive book, over 1200 pages with 1500 color images. There are 70 expert authors that contributed chapters to this work.

\* Manual of Reptiles. Girling, S and Raiti, P. British Small Animal Veterinary Association. Worthing, West Sussex, 2004.

\* Biology, Husbandry and Medicine of the Green Iguana. Jacobson, E. Kreiger Publishing Co, Malabar, FL. 2003.

