ANESTHESIA IN REPTILES

Part 2: inhalation anesthesia

T. Bouts, F. Gasthuys

Department of Surgery and Anesthesiology of Domestic Animals
Faculty of Veterinary Medicine, Ghent University
Salisburylaan 133, B-9820 Merelbeke, Belgium
tom.bouts@rug.ac.be

ABSTRACT

The first part of the article provided a review of injection anesthesia in reptiles. In the second part, inhalation anesthesia in these species is highlighted. Specific attention is given to the anatomy and physiology of the respiration system in reptiles and the pre-anesthetic preparation. Practical details of the equipment, endotracheal intubation and monitoring are provided.

Problems such as breath-holding can occur in reptiles. Another problem is the enormous variability in species and size. In snakes it is common practice to intubate the animal awake and "gas" it down. This technique is neither justified nor possible in venomous snakes. Induction of anesthesia in these animals can be done using different injection techniques (see Part 1) or the box induction. Intubation can be done once the animal is asleep. Lizards have an easily accessible tail vein, while the jugular vein can be used for injection in cooperative chelonians. Propofol is the product of choice for anesthetic induction in these species (see Part 1). However, as in non-venomous snakes, "conscious" intubation is becoming more and more popular in lizards and chelonians.

Isoflurane is the most widely used volatile anesthetic in reptiles, although satisfactory results have been reported using sevoflurane in exotic practices in the UK and USA over the last two years.

SAMENVATTING

In het eerste deel van dit artikel werd een overzicht gegeven van de injectieanesthesie bij reptielen. Het tweede deel omvat de verschillende praktische kanten van de inhalatieanesthesie bij deze dieren. Eerst worden de specifieke anatomie en fysiologie van het ademhalingssysteem samen met het pre-anesthetisch onderzoek belicht. Vervolgens worden praktische gegevens omtrent de uitrusting, de intubatie en monitoring besproken.

Bepaalde reptielen kunnen het fenomeen van "breath holding" vertonen. Een bijkomend probleem is de enorme variatie tussen de verschillende species. Slangen kunnen meestal geïntubeerd worden in een wakkere toestand om ze vervolgens met volatiele gasmengsels te induceren. Deze techniek is niet mogelijk en niet verantwoord bij agressieve of giftige dieren. Hier is een inductie door middel van injectie vereist (zie Deel 1) of kan de boxinductie toegepast worden. Hagedissen hebben meestal een makkelijk toegankelijke staartvene, terwijl de *V. jugularis* gebruikt kan worden bij coöperatieve schildpadden. Propofol is aangewezen bij deze species om een inductie van de anesthesie te bekomen. Het intuberen van bewuste hagedissen en schildpadden wordt meer en meer toegepast.

Isoflurane is het meest gebruikte volatiele anestheticum voor reptielen. De laatste twee jaar werden goede resultaten vermeld met sevoflurane in verschilende gespecialiseerde praktijken in de Verenigde Staten en het Verenigd Koninkrijk.

INTRODUCTION

The numbers of reptiles being referred for consultation in small animal practice has been increasing in recent years. The different methods for administering injection anesthesia were reviewed in the first part of this article (Bouts and Gasthuys, 2002).

Volatile anesthetics can also be used for anesthesia of reptiles. Inhalation anesthesia has several advantages over injection anesthesia. It is relatively safe and easy to use. The depth of anesthesia can be more accurately controlled and recovery is much faster. The major disadvantages are the financial impact of the

equipment and the pollution of the environment when no adequate scavenging is provided (Bennet *et al.*, 1999).

This article will discuss the respiratory anatomy and physiology of reptiles, the anesthetic gases and equipment used with them, and the possibilities for monitoring these procedures.

ANATOMY AND PHYSIOLOGY

The **larynx** of reptiles is located rostrally in the mouth at the base of the tongue. In snakes the glottis is located in the most cranial part of the mouth. Chelonians have a thick, fleshy tongue, which makes the localization of the glottis sometimes problematic. The use of an adapted small-sized laryngoscope can be of help to visualize the different intra-oral structures (Burke, 1978; Bennet,1996).

The **trachea** of the squamata is composed of incomplete tracheal rings with a dorsal tracheal membrane. Many snakes have a so called tracheal lung (alveolar tissue on the dorsal surface of the trachea), which allows them to breath, even after ingestion of larger preys which compress the abdominal lungs. The trachea of chelonians has complete rings and is very short. The positioning of the endotracheal tube in these animals is not without danger: damage to the mucosa of the trachea and "one lung" intubation have been described (Burke, 1978; Bennet, 1991). The **lungs** of most reptiles are the so-called endothelium-lined sacs. The lungs have ridges on their surface, forming a reticulated pattern and increasing the surface area. In more developed species there are foldings of the lung wall, which further increase the surface area. In most snakes, the left lung is vestigial or absent. However, pythons and boas have better developed left lungs. The right lung of snakes extends caudally, and can approach the cloaca in some species. At this level it loses the reticular pattern and becomes the air sac. The lungs in chelonians are situated dorsally of the viscera. When these animals are positioned in dorsal recumbency, the viscera compress the lungs and reduce their tidal volume, making artificial ventilation necessary. The lungs of reptiles are considered fragile so care must be taken when positive pressure ventilation is applied in these animals (Davies, 1981; Bennet, 1991).

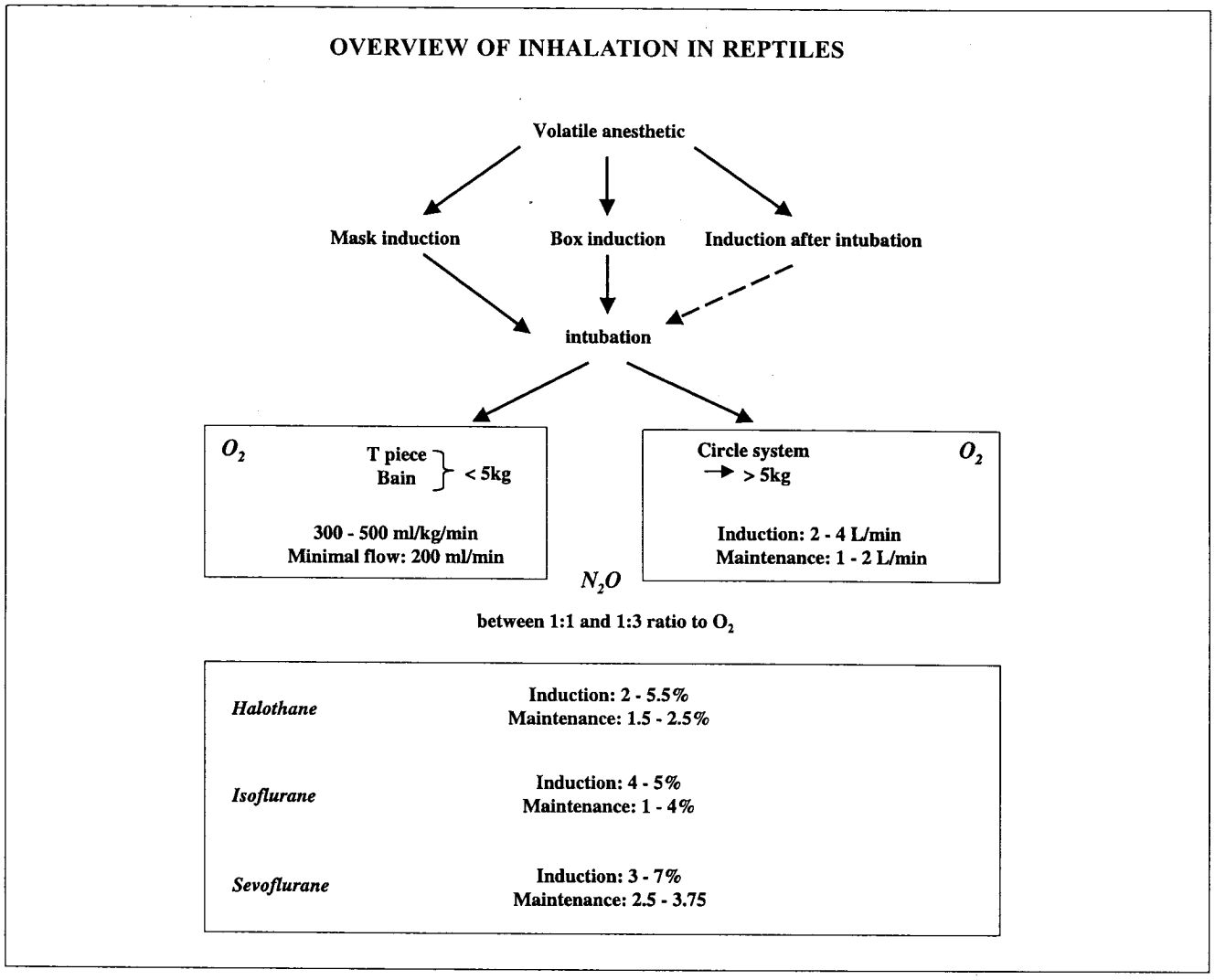
Reptiles do not have a functional muscular diaphragm but rather a combined pleuroperitoneum or coelomic cavity. On the other hand, chelonians, crocodiles and iguanas have a membranous structure between the abdomen and the thorax, separating the thoracic from the abdominal viscera (Burke, 1978; Bennet, 1991).

Reptiles normally breathe through the nose but they are also capable of breathing through the mouth. The negative pressure required for the movement of air into the lungs is generated by two different mechanisms. Snakes and lizards use the intercostal muscles and the muscles of the trunk and abdomen to create negative pressure. The walls of the lungs contain smooth muscles which contract and relax actively to direct air through the lungs (Davies, 1981). The first mechanism can stop at deep levels of anesthesia because the muscles become paralyzed, making it necessary to use artificial respiration. Both mechanisms make it possible to breath, even when there is an open coelomic cavity, for example due to abdominal surgery. In contrast, chelonians have no intercostal movements. In these species, pressure differences are generated by moving the viscera, limbs and pelvic girdle, which causes the abdominal muscles to contract, thus compressing the organs of the visceral cavity (Bennet, 1991). Many reptiles are able to hold their breath, which changes the aerobic metabolism into anaerobic metabolism. Turtles from the genus *Pseudemys* were reported to survive up to 27 hours in a 100% nitrogen environment (Calderwood, 1971; Schildger *et al.*, 1993). Green iguanas (*Iguana iguana*) can hold their breath up to 4.5 hours (Bennet, 1991). The possibility of holding their breath makes gas induction by mask or box in these animals almost impossible.

PRE-ANESTHETIC PREPARATION

Pre-anesthetic starvation in reptiles is useful for several reasons. The main reason for starvation is not to prevent regurgitation, since the reptile glottis is normally closed between breaths. Rather, it functions mainly to prevent compression of the lungs due to the earlier digestion of large meals (Bennet, 1991). Another reason is that coelioscopy is easier to perform when there is no food in the viscera (Schildger *et al.*, 1993). The periodof starvation differs from species to species. Eighteen hours is recommended in smaller lizards and chelonians, and 72 to 96 hours in snakes, larger herbivore lizards and tortoises (Malley, 1998).

A simple clinical examination can be performed in most reptiles. It is of major importance to check the mouth for *stomatitis ulcerosa* because this could make intubation impossible (Schildger *et al.*, 1993). Depending on the anamnesis and the external clinical



signs, diagnostic samples can be taken and blood can be sampled for blood cell count and biochemistry (Bennet *et al.*, 1999).

ANESTHETIC CARRIER GASES

Volatile anesthetics require carrier gases to enable the passage of these agents from the anesthetic equipment to the animals. Several gases can be used for this purpose.

Oxygen (O_2) transports the volatile anesthetic into the lungs, and prevents and corrects potential hypoxemia (Frye, 1981). The flow rate required to provide a sufficient oxygen supply in a non-rebreathing system is approximately twice the minute volume, which is thought to be between 300 and 500 ml/kg/min in reptiles. In the rebreathing systems, including the classic circle system, a flow of 2 to 4 L/min of oxygen is advised for induction while 1 to 2 L/min of oxygen can be used safely for maintenance of anesthesia (Bennet, 1991). Pure oxygen usually induces apnea in reptiles (see further). Oxygen can be mixed with compressed air to reduce the inspiratory percentage of oxygen.

Nitrous oxide (N_2O) can be included as part of the fresh gases in order to speed up induction but mainly to provide a more adequate level of analgesia (Malley, 1998; Heard, 2001). It also reduces the required amount of volatile anesthetic and provides some muscle relaxation (Frye, 1981). Nitrous oxide can be supplied at between a 1:1 and a 1:3 ratio compared to oxygen. Disadvantages include the possibility of diffusion hypoxia at the end of anesthesia (nitrous oxide diluting the oxygen in the lungs by its massive release from the blood) and the ability to expand closed spaces filled with air in the body (exchange with nitrogen) (Malley, 1998). It is recommended that the supply of to reduce the amount of nitrous oxide or to disconnect it entirely after induction of anesthesia (Frye, 1981).

Carbon dioxide (CO_2) can be administered as a respiratory stimulant, especially in breath holding species. When inducing turtles of the *Pseudemys* genus, 5 to 10 % CO_2 can be mixed with the other carrier gases in order to increase ventilation rate and minute volume and so speeding up induction (Jones, 1977; Frye, 1981; Bennet 1991). However, Millichamp

(1988) reported that respiration in reptiles is more regulated by hypoxia rather than by hypercapnia so the use of using carbon dioxide can be considered.

VOLATILE ANESTHETICS

In reptile anesthesia several volatile anesthetics can be used, such as halothane, isoflurane and sevoflurane. Ether, enflurane and methoxyflurane are out-of-date and are presently not justified for use in all animals. Inhalation anesthetics act by producing an adequate partial pressure of anesthetic in the brain to induce a desired level of anesthesia (Steffey, 1996).

Halothane is a relatively old fluorinated organic inhalation agent. It is non-flammable and non-irritating to respiratory tissues. This agents makes the myocard more sensitive for catecholamine induced arrhythmias, produces a dose-dependant cardiopulmonary depression and has also been reported to cause liver damage (Frye, 1981; Bennet, 1991). The latter finding is certainly a matter of discussion since repetitive anesthesia using halothane in different animals has failed to confirm this reported hepatic side effect. Halothane can decompose due to exposure to light so a preservative, thymol, is added. This preservative forms a sticky deposit, which causes technical problems in the vaporizer. (Steffey, 1996).

An old and simple method for the administration of halothane to poisonous snakes and lizards involved placing a halothane-soaked pad in a box. The approximate dose is 5 ml/2840 cm^3 (1ft^3) of box space. This usually produced the onset of anesthesia in 5 to 33 minutes and an effective duration of anesthesia of 5 to 20 minutes (Jones, 1977). Since halothane is heavier than air, it remains at the bottom of the box; snakes have been reported to try to "escape" from the anesthetic vapors by raising their heads above the gas level (Bennet, 1991).

Precision vaporizers are presently used for the administration of volatile anesthetics. When using halothane, vaporiser settings of 2 to 5% are recommended for induction and 1.5 to 2.5% for maintenance (Bennet, 1996). Breath-holding species can develop long-lasting periods of apnea because of the specific odor of halothane. An alternative is to start with 100% oxygen and gradually increase the halothane concentration. During induction an excitement phase can be observed prior to anesthesia but this phenomenon was not reported to occur during the recovery phase (Bennet, 1991). Recovery after halothane anesthesia is usually complete after about one hour (Jones, 1977).

Isoflurane has similar dose-dependant cardiovascular properties as halothane but it is almost completely eliminated by the lungs (99.8 %), making it the agent of choice for debilitated patients (Bennet, 1996). It should be noted that isoflurane depresses the blood pressure and the ventilation and is a poor analgesic (Malley, 1998). Isoflurane is certainly more expensive than halothane. When working with a vaporizer, a 4 to 5% concentration can be used for induction and a 1 to 4% concentration is suitable for maintenance of anesthesia. Induction occurs in 6 to 20 minutes and recovery is usually complete within 30 to 60 minutes (Bennet, 1996). However, longer periods of recovery have been reported

A recently developed volatile anesthetic in reptile medicine is **sevoflurane**. This fast-acting anesthetic is still relatively expensive (2 times the cost of isoflurane), while it possesses only half the tissue and blood solubility of isoflurane. Overall sevoflurane is about four times as expensive as isoflurane (Heard, 2001). This agent has already been tested in desert tortoises *(Gopherus agassizii),* with an induction time of approximately 3 minutes and a recovery time of approximately 28 minutes. Blood pressure dropped but heart rates remained normal during anesthesia. The induction concentration was 3 to 7% and the maintenance concentration was 2.5 to 3.75% (Rooney *et al.*, 1999). Further research is still required in other species to evaluate the properties of sevoflurane in reptiles.

EQUIPMENT

A relatively simple technique for inducing anesthesia is the induction **box** (usually a simple construction in plastic or glass, with an inlet and an outlet for the anesthetic gases) in which the reptile is placed. Two methods of inducing anesthesia have been described. A cotton wool pad is soaked in a volatile anesthetic and dropped into the box. This is probably the easiest method, but it can induce several dangerous complications, including overdose. The concentration of the anesthetic vapor depends on several factors, such as the size and temperature of the box, the volume displaced and the anesthetic uptake by the reptile (Calderwood, 1971; Jones, 1977). It is safer to use a precision vaporizer that delivers gases in small volumes into the box, so better control of the amount of vapor can be achieved (Calderwood, 1971).

Another method for inducing anesthesia is mask induction. Several **masks** are commercially available, mostly for small animals (see Figure 1), but these are

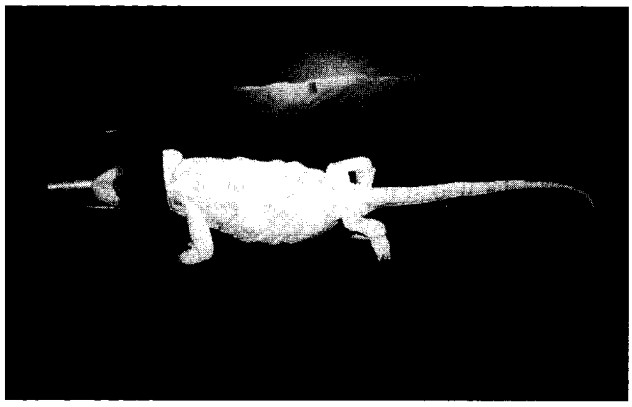


Figure 1. Mask induction (commercial small animal mask) in a chameleon with egg retention.

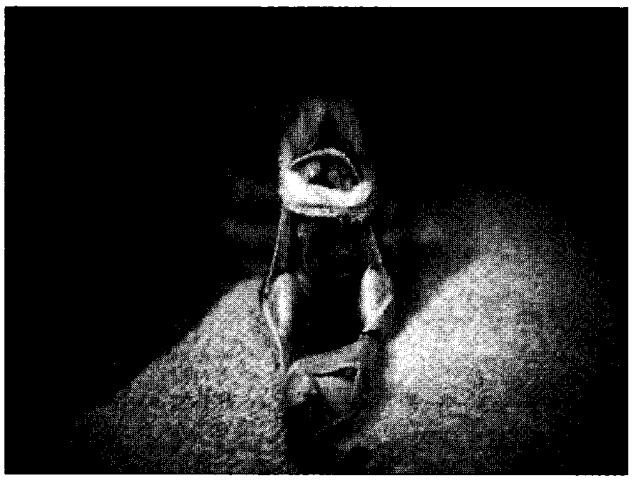


Figure 3. Intubated snake; the tracheotube (3 mm ID) is taped on the lower jaw of the animal.

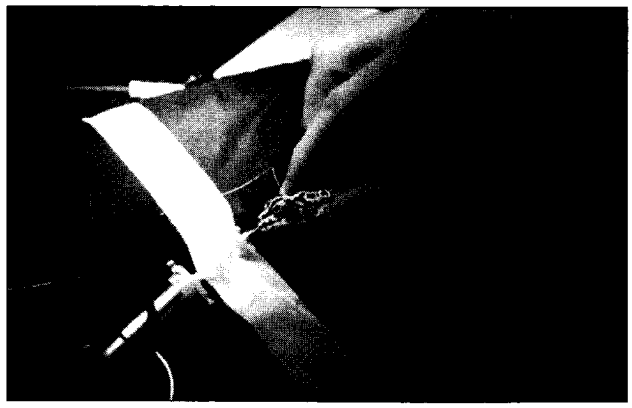


Figure 2. Intubation of Leopard Gecko *(Eublepharius macularius)* using the shaft of a 22 G intravenous catheter and a commercial Ayre's T-piece.

often too big for small reptiles. In most reptiles homemade masks can be produced from all kinds of materials such as plastic bottles and syringes. The mask is placed over the head and anesthetic gases are provided until a tracheotube can be placed (Calderwood, 1971). Specially designed masks including a scaving system are presently being distributed, mainly for use in experimental animals; this system can certainly be applied with most reptiles (Fluovac System, International Market Supply, Cheshire, UK)

In easy to handle snakes, a **tracheotube** can be placed in the awake animal without specific problems. The commercial tracheotubes often have a diameter which is too large for reptiles. Again, homemade tracheotubes can be constructed using urinary and intravenous catheters. The tracheotube should be secured to the rostrum to avoid its dislodgment during patient handling (see Figure 2 and 3) (Frye, 1981).

The **anesthetic machine** supplies a mixture of different gases to the breathing system. Medical gas sources (small compressed gas cylinders or central gas supply for O_2 and N_2O), a pressure regulator, a flowmeter for each gas and a precision vaporizer for each volatile anesthetic are essential parts of this machine. Most vaporizers require a minimum flow of more than 200 mL/min. Adequate scavenging systems are required to avoid pollution and possible health risks for the users. (Hartsfield, 1996).

The **breathing system** delivers the anesthetic gases and volatile anesthetics to the patient, removes CO_2 from the exhaled gases, and can provide support for manual ventilation (Hartsfield, 1996). The choice of breathing systems depends mainly on the body weight. Reptiles weighing less than 5 kg require a non-rebreathing system (T-piece or coaxial) because of the lower resistance and the minimal rebreathing of expired CO_2 (see Figure 2). If a non-rebreathing system were used in larger reptiles, the flow rate would be too high to prevent rebreathing (expensive); therefore these animals usually require a classic circle system (internal diameter 22 m) (Calderwood, 1971; Bennet, 1991).

It is highly recommended that reptiles should be ventilated (mostly intermittent positive pressure ventilation) during anesthesia because pure oxygen (100%) usually induces a relatively long period of apnea. This can wake up the animals (Bennet, 1991). Artificial respiration can be done manually by squeezing the rebreathing bag, though a pediatric **respirator** can also be used for this purpose. The normal respiratory rate for most reptiles is 10 to 20 breaths/min, but when pure oxygen is used, 4 to 6 breaths/min is adequate. Because of the fragility of the lungs, it is recommended not to apply a pressure of more than 12 cm H_2O (Burke, 1978; Bennet, 1991; Bennet, 1996).

ENDOTRACHEAL INTUBATION

It is important to have good a knowledge of respiratory anatomy and physiology in reptiles because problems can arise, such as one lung intubation in chelonians (see above). To open the mouth in reptiles, a gentle ventral traction on the lower jaw can be applied. In aggressive reptiles that are awake, tapping on the rostrum may open the mouth. However, the manual opening of the mouth of aggressive reptiles and larger tortoises and lizards (sharp beaks) can induce dangerous situations for the anesthesiologist. When the mouth is open, a mouth gag should be placed to prevent closure. Lidocaine gel or spray (2 %) can be sprayed on the glottis (possible overdose in smaller body weight). Once the airway is visible, the tube is inserted when the glottis opens. Finally, the tube is taped to the upper or lower jaw of the reptile, thus assuring adequate fixation during anesthesia (Heard, 2001) (see Figure 3). The use of endotracheal tubes with inflation of the cuff is contraindicated in reptiles with close tracheal rings.

MONITORING

As in mammals, four stages of anesthesia have been described in reptiles. Stage I is characterized by slow voluntary movements, no muscle relaxation and a positive righting reflex (i.e. when the reptile is placed on his back, it tries to roll back). Painful stimuli are still experienced. Stage II involves very few spontaneous movements, moderate muscle relaxation and a poor righting reflex. The responses to painful stimuli are either reduced or absent. Stage III is the surgical level of anesthesia, involving no movement, the absence of the righting reflex and no responses to painful stimuli. Finally, stage IV is the toxic level of anesthesia approaching death (Malley, 1998). Several reflexes can be used to monitor **anesthetic depth** such as the righting reflex, the tail, tongue or foot withdrawal reflexes (withdrawal of this part of the body when gently squeezed) and the corneal reflex. Surgical anesthesia is present when the righting, tail and foot withdrawal reflexes are lost. Anesthesia is too deep when the corneal reflex or tongue withdrawal reflex is lost (Bennet, 1991; Schildger *et al.*, 1993; Malley, 1998). It is useful to know that snakes relax from head to tail during inhalation anesthesia and recover in the opposite direction (Heard, 2001).

Reptiles are ectotherms and depend mainly on external heat sources for their **thermoregulation**. Due to this characteristic, the body temperature must be maintained within the preferred temperature ranges (see Part 1) not only during anesthesia but also in the recovery period. Warm water or air blankets and heat lamps are useful alternatives for this purpose. Special attention must be given to preventing local overheating when using simple infrared lamps (little or no increase in cloacal temperature). Although prevention of hypothermia is important during anesthesia, hyperthermia is even more dangerous. The body temperature can easily be monitored with a cloacal commercial temperature probe. (Schildger *et al.*, 1993; Heard 2001). The specific data of the preferred body temperature of each species can be used as a guideline for the monitoring of the thermoregulation. A temperature of 30 °C usually induces severe hypothermia in heliothermic reptiles.

Monitoring of the heart rate in reptiles is often not possible using the classic external auscultation techniques. A small-sized **esophageal stethoscope** and/or **ECG** can be applied. In snakes the right leg (RL) and left leg (LL) electrodes are placed on either side of the vent while the right arm (RA) and left arm (LA) electrodes are positioned midway in the cervical region (Frye, 1995). In lizards and chelonians the electrodes are placed on the conventional places as in dogs and cats (Heard, 2001). Frye (1995) developed a three-lead esophageal ECG for use in snakes, with satisfactory results. QRS waves of reptiles are generally inverted and slurred but the information about arrhythmias and blocks is certainly of value (Bennet, 1996; Heard, 2001).

An adapted **Doppler blood flow monitor** (Vettex, Huntleigh Diagnostics, Cardiff, UK) with an 8 Mhz flat or pencil probe or a 5 Mhz flat probe can used to detect the sound of blood flowing in the heart or large blood vessels. The frequencies of transmitted sound waves are altered when these waves are reflected by the moving red blood cells. This equipment is of great value in smaller reptiles and chelonians on which it is difficult to place an ECG or listen to the heart with a classic stethoscope. A weak signal can be due either to a decrease in blood flow or to a malpositioning of the probe. In chelonians the detector is placed either directly on the plastral surface or on the soft surface covering the region between the medial aspect of one forelimb and the neck. In lizards the probe is placed on the midline cranial to the forelimbs, and in snakes the probe is placed in the ventral midline approximately 15% down the length of the snake and moved caudally until the heart sound is heard (Frye, 1995, Heart, 2001).

Pulse oximetry provides information on the pulse rate and peripheral hemoglobin saturation (SpO_2).

This technique has become popular for monitoring anesthetized reptiles. Two pulse oximeter probes (POP's) have been developed. Transmission POP's measure signals across perfused tissue, while reflectance POP's use two different wavelengths. The transmission POP's (such as the classical human finger probes) can be placed on the tongue, while the reflectance POP's are positioned in the esophagus or the rectum. Reflectance pulse oximeters have been reported to give more reliable results in reptiles (Bennet, 1996; Malley, 1998; Heard, 2001). Problems with the transmission pulse oximeter probes can be encountered in reptiles with darkly pigmented skin (little penetration of the emitted light). The upper range of the heart rate of the pulse oximeter must be relatively high (up to 250 beats per minutes) in order to be useful in smaller reptiles.

Capnometry is used to monitor respiratory rate and CO_2 concentrations in inspired and expired gases in mammals. Because the three-chambered heart induces cardiac shunting in reptiles, the end tidal CO_2 does not reflect the arterial CO_2, thus making capnography less accurate in these animals (Heard, 2001).

RECOVERY

The administration of volatile anesthetic should be terminated 15 to 20 minutes before the end of surgery to hasten recovery (Heard, 2001). Recovery should occur in a quiet, stress-free environment with temperature and humidity in the preferred optimum range for the species (Bennet, 1996). The endotracheal tube must be kept in place until the reptile respires spontaneously. If necessary, artificial respiration should be applied at least once per minute (Bennet, 1996). It is better to use room air (21 %) than pure oxygen (100 %) because respiration in reptiles is stimulated more by a low oxygen pressure rather than by a low carbon dioxide pressure (Bennet *et al.*, 1999). If necessary, the return from artificial ventilation to spontaneous respiration can be facilitated by the administration of doxapram (5 mg/kg IV or IC) (Malley, 1998).

Aquatic reptiles should not be allowed to swim until recovery is complete (Schildger *et al.*, 1993).

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