

CHAPTER

# 39

ANESTHESIOLOGY

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**W**ith the refractory attitudes of many birds toward even mild restraint, sedatives and local anesthetics are of little use in most avian species. General anesthesia, however, with appropriate agents, can enable clinicians to safely and rapidly perform fluid administration, emergency procedures, blood collection and radiography, or to perform prolonged invasive surgical procedures in avian patients.

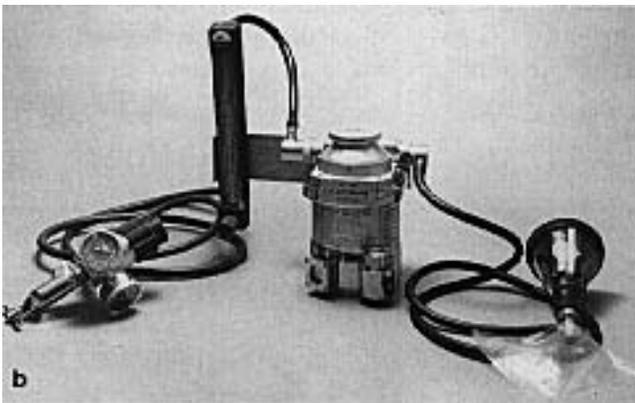
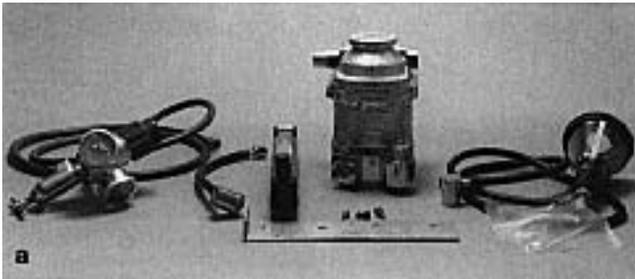
Historically, avian anesthesia has been a problem fraught with continuous debate. Many clinicians have had their preferred drug “cocktails,” and there were many conflicting views with regard to dosage ranges and choice of anesthetic regime. Anesthesia machines have been altered in an attempt to meet the specialized needs of avian patients, and numerous modified endotracheal tubes, non-rebreathing bags and delivery systems have been implemented. Sophisticated monitoring systems and equipment designed for or found to be suitable for avian patients are now commercially available.

As in other animal species, general anesthesia in birds can be accomplished with either injectable or inhalant anesthetic agents. The goal of anesthetizing a patient is to select the safest drug that allows the minimum amount of physiologic changes. (The reader is referred to Section Seven for use of anesthetics in non-psittacine species.) Injectable anesthetics are far inferior to gas anesthetics for use in avian patients, and for private avian practice, isoflurane is the only recommended anesthetic. This is particularly true given that the vast majority of patients are anything but healthy and are less tolerant of the physiologic compromises induced by most injectable and other inhalant anesthetics.

The basic principles of risk assessment and patient support used for mammalian anesthesia are also applicable to the avian patient. Ability to assess the condition of avian patients has improved, as has the ability to provide physiologic support during the anesthetic episode.

## Anesthetic Agents and Equipment

The ideal avian anesthetic agent is one that creates minimal stress in administration, has a high therapeutic index, provides for rapid induction and recovery, induces minimal physiologic changes, provides adequate restraint for the desired procedure and can be safely used in critical cases. Contraindications for anesthetizing an avian patient should include severe obesity, fatty liver, liver or kidney failure, dehydration, shock, anemia, dyspnea and fluid in the crop. Unfortunately, patients presented with many of these problems are those that require anesthesia for proper resolution of the case. The choice of anesthetic agent must be based on the patient's status and the working conditions that the clinician faces (eg, field vs. hospital anesthesia). In all situations, the anesthetic of choice is isoflurane.<sup>a,b</sup> There are some indications for the use of injectables in the field, and recent work with reversal agents may make the use of injectables more appealing to some avian practitio-



**FIG 39.1** An isoflurane anesthesia circuit for use in birds that will fit into a small tool box is commercially available. **a)** The components of the system are shown separated and **b)** connected to form a functional circuit (courtesy of Exotic Animal Medical Products).

ners. However, an isoflurane anesthetic unit designed for field use will fit into a small tool box (10" x 12" x 20"); the only other necessary equipment is an oxygen source (Figure 39.1).

When compared to injectable anesthetics, inhalation agents have numerous advantages. They can be titrated to effect, have a more consistent therapeutic index and provide for rapid induction and smooth, rapid recoveries. Additionally, the anesthetic episode can be maintained for variable durations as dictated by the procedure and, particularly with isoflurane, effects can be instantly reversed.

### Physiologic Effects of Inhalant Anesthesia

In administering avian inhalant anesthetics, there are several important differences between the mammalian and avian respiratory system that should be addressed. The paramount difference is that the avian lung does not have alveoli. Instead, the air capillaries function as the anatomic location of gas exchange.<sup>28</sup> Avian species also lack a diaphragm, and inspiration is totally dependent on the correlative movement of the coracoids, ribs and sternum. The total lung capacity of avian species is much less than that of an equivalent-sized mammal; however, due to the air sac system, the total respiratory volume is substantially greater. There is also a high gas exchange surface-to-volume ratio that accounts for more efficient gas exchange. This efficiency accounts for the rapid equilibration of inspired components with arterial blood and for the rapid induction, rapid changes in depth of anesthesia and speed of recovery when inhalant anesthetics are used in birds. Recovery from isoflurane is primarily a function of the excretion of the gas by the lungs. Full recovery with agents such as methoxyflurane that are highly metabolized depends on the biotransformation of the agent by the liver. Birds are very sensitive to CO<sub>2</sub> concentrations, and if the blood is depleted of CO<sub>2</sub>, the patient will become acutely apneic. The minimum level of CO<sub>2</sub> in the blood that is necessary to stimulate respiration in birds has been suggested to be 25 torrs.<sup>6</sup>

Because of the anatomy and structure of the avian respiratory system, even healthy birds may not be properly oxygenated when anesthetized and placed in dorsal recumbency.<sup>6</sup> It may be impossible for some species that have a large pectoral muscle mass (eg, Galliformes and Anseriformes) to adequately ventilate. Because of their unique respiratory anatomy, intubation and the use of gentle intermittent positive

pressure ventilation (IPPV) (20 to 40 per minute at 15 mm H<sub>2</sub>O) is strongly recommended in anesthetized patients.<sup>28</sup>

### Isoflurane

Isoflurane is rapidly replacing halothane and methoxyflurane as the gas anesthetic of choice for small animal patients. This surge in popularity is based on isoflurane's rapid induction time, rapid and smooth recoveries, rapid change in anesthetic level, high margin of safety for both the patient and hospital staff, reduced arrhythmogenic properties, reduced cardiovascular depression and reduced respiratory depression. The drug can also be safely used to obtain diagnostic information from high-risk and critically ill birds. Recovery from even long surgical procedures requires only minutes.

Isoflurane can have a dose-related depressant effect on the respiratory and cardiovascular system. Fortunately, there is a substantial interval between respiratory and cardiac arrest.<sup>1,9,11,27</sup> The hypotensive effects of isoflurane have been shown to be severe in cranes. The effect was dose-dependent and was potentiated by spontaneous respiration when compared to assisted ventilation (IPPV).<sup>19</sup> Isoflurane is only 0.3% metabolized compared to 15% for halothane and 50% for methoxyflurane; thus, isoflurane does not produce the hepatic damage induced by halothane and methoxyflurane. However, it has been evaluated in the field for only 12 years, and precautions should still be used with this anesthetic gas. The low metabolism rate also means that when the gas is expired by the lungs, few residues or toxic metabolites remain to further hinder the patient's recovery. Isoflurane has a blood solubility of 1.4 compared to 13 for methoxyflurane and 2.4 for halothane. Because the solubility is so low, the agent is not dissolved in the blood, and the speed of induction and recovery are extremely rapid. Minimum alveolar concentration (MAC) has been found to be approximately 1.3 (cranes and ducks).<sup>18,19</sup> This would indicate that higher settings for isoflurane delivery might predispose the patient to apnea, cardiac arrhythmias and cardiac arrest.

### Methoxyflurane

Methoxyflurane is 50% metabolized, has a high degree of organ toxicity and produces prolonged effects on physiologic parameters. This gas is highly soluble in blood, which accounts for its relatively long periods of induction and recovery (may take hours). This solubility prevents the rapid change in anesthetic depth, which makes the anesthetized bird easier to

maintain. However, the quantity of gas dissolved in the blood also prolongs the time required to lighten a bird in critical situations. Methoxyflurane is known to induce major hepatic and renal dysfunction in chronically exposed hospital personnel, and scavenging systems should be used to remove waste gas from the hospital setting. Because of methoxyflurane's low volatility, techniques have been described for administering the gas in open anesthetic systems. This is an extremely dangerous method of providing anesthesia in birds, and if an adequate oxygen supply is not assured, it will result in rapid death of the patient.

### Halothane

Halothane is a relatively nonirritating gas that requires a precision vaporizer for proper delivery. The blood:gas coefficient for mammals is 2.3, which, when combined with the rapid arterial gas exchange that occurs in the avian respiratory system, accounts for a rapid induction and recovery. Induction is usually achieved within two to five minutes at a two to three percent level; recovery time depends on the length of the procedure but generally varies from 5 to 20 minutes. Halothane will induce a rapid decrease in the heart rate that returns to normal shortly after ceasing anesthetic administration. Maintenance is usually achieved between 1 to 1.5%, which decreases as the length of the procedure increases, and decreases with the degree of induced hypothermia. Halothane is only 15% metabolized. Disadvantages of halothane in the avian patient are that 1) apnea and cardiac arrest often occur at the same time, and 2) the gas does sensitize the heart to catecholamines, which may induce arrhythmias, particularly with longer surgical procedures. The agent can cause liver disease in chronically exposed hospital personnel, and scavenging systems should be used to remove waste gas from the hospital setting. Recoveries with halothane are more prolonged than with isoflurane.

## CLINICAL APPLICATIONS

**Isoflurane is an ideal anesthetic in birds because of its:**

- High therapeutic index
- Rapid induction and recovery
- Minimal physiologic changes
- Adequate restraint for many procedures
- Safety in critical patients
- Reduced toxicity

### Oxygen

The oxygen flow should be high enough to ensure that a precision vaporizer is accurate in its delivery of the anesthetic gas. For most precision vaporizers, the minimum flow rate is 500 ml/min. Some vaporizers function adequately at low settings but the manufacturer's recommendations should always be followed. If a semi-open system is used, the oxygen flow should be three times the respiratory minute volume, which for a 450 g bird is about 275 ml/min. As a general guideline, this ratio can be used to determine the respiratory minute volume of any avian species. For most psittacine birds, the oxygen flow rate during induction is 1 l/min and maintenance is 0.5 to 1 l/min depending on the size of the patient.

### Nitrous Oxide

Nitrous oxide (N<sub>2</sub>O) has successfully been used in birds in combination with isoflurane anesthesia. N<sub>2</sub>O is not potent enough to induce anesthesia on its own; however, it does allow for the reduction in the percentage of isoflurane necessary for anesthetic maintenance. Because cardiovascular and respiratory depression caused by isoflurane are dose-dependent, N<sub>2</sub>O is an important addition to the anesthetic regime. N<sub>2</sub>O does have the characteristic of diffusing into closed gas spaces faster than nitrogen (room air) can diffuse out. This means that N<sub>2</sub>O is contraindicated in situations where dead gas spaces are present. Because the avian respiratory system including the air sacs freely intercommunicate, the use of N<sub>2</sub>O is not contraindicated. Some species differences do exist. For instance, diving birds have naturally occurring subcutaneous air pockets, and the use of N<sub>2</sub>O in these birds may lead to subcutaneous emphysema.<sup>3</sup>

### Pre-anesthetics

The routine use of atropine as a pre-anesthetic has been avoided in avian patients because it thickens respiratory secretions, slows gastrointestinal motility and increases the heart rate. The thickening of respiratory secretions could contribute to a life-threatening occlusion in patients intubated with small-diameter endotracheal tubes. Additional elevation of the heart rate is not desirable in patients that already have a rapid rate. Glycopyrrolate does not have as marked effect on the heart, but it too causes thickening of respiratory secretions. Consequently, these drugs are used only as specific therapy for bradycardia. Further studies on the use of these drugs in birds are needed.

### Injectable Anesthetics

Injectable anesthetics in birds have the same disadvantages that are recognized in mammalian species. There is a tremendous variability in therapeutic dosages and physiologic effects, both at the species and individual patient levels. Likewise, many injectable anesthetics do not provide an adequate plane of anesthesia without reaching tissue levels that threaten the life of the patient. There is minimal ability to titrate injectable agents to effect, so levels of anesthesia may be insufficient to perform a procedure or may be so deep that the patient is in danger. With most commonly used injectable agents, the anesthetic level cannot be rapidly decreased, and the recovery is prolonged because the drug must be totally removed by metabolic pathways. Increased recovery times create excessive stress, increase the period of hypothermia and prolong the deviation from a physiologically normal state. With any anesthetic episode, a major goal should be to minimize the time between induction and recovery, and injectable anesthetics do not effectively meet this criteria. Unfortunately, when using parenteral anesthetics, the period of recovery may be far longer than the duration of useful anesthesia.

The most commonly reported injectable anesthetics used in birds are combinations of ketamine and xylazine and, less frequently, ketamine and diazepam. Etorphine, methoxymol, propafol, midazolam, tiletamine/zolezapam and barbiturates have all been used in birds.<sup>4,7,12,15</sup> Initial data are promising for some of these drugs, but in many cases actual clinical trials are not available. In general, phenobarbital, methohexital, thiobarbiturates and barbital should not be used in companion birds. They have a low margin of safety, produce prolonged violent recoveries, must be given IV to prevent perivascular damage and at best provide radically variable levels of anesthesia.

Ketamine, a cyclohexamine, produces a cataleptic state that inhibits movement, but does not provide adequate analgesia for major surgical procedures. This drug has a highly variable effect in different avian species. It is metabolized by the kidneys and is therefore contraindicated in patients with renal insufficiency. Dose ranges for various species are reported from 5 to 75 mg/kg. The drug is most commonly administered IM, and the first signs of incoordination occur within three to five minutes. The typical duration of anesthesia is 10 to 30 minutes, and recovery may take from 30 minutes to several hours, which is completely dose-dependent.

Ketamine anesthesia is typified by cardiac and respiratory depression, increased blood pressure, reduced body temperature, slow violent recoveries and prolonged physiologic changes.

Ketamine is rarely used alone. Because of the muscle rigidity produced by this drug and the inadequacy of the analgesia achieved, ketamine is most often used in combination with either xylazine or diazepam.<sup>12,20</sup> The ratio for both ketamine/diazepam or ketamine/xylazine combination is 10:1 on a mg/kg basis. Either combination provides for more rapid induction, smoother maintenance and less violent recovery than when ketamine is used alone.

Xylazine produces good muscle relaxation and transient analgesia. It can cause bradycardia and heart blocks.<sup>20</sup>

Diazepam is an excellent sedative and provides some muscle relaxation.<sup>20</sup> Ketamine/diazepam combinations can be useful when mild restraint is required.

The dosages for the drugs to be administered in combination are calculated based on a ketamine dosage of 5 to 30 mg/kg IM (or 2.5 mg to 5.0 mg IV) mixed with xylazine 1.0 mg to 4.0 mg IM (or 0.25 to 0.50 mg IV). Alternatively, diazepam (0.5 mg to 2.0 mg/kg IM or IV) may be substituted for the xylazine.<sup>2,4,5,8,12,28</sup> The two drugs chosen are mixed together and administered either intramuscularly or intravenously. Reduced doses are necessary in seriously ill, pediatric and geriatric patients and for intravenous administration. Ratites require only 3 mg/kg of ketamine. Because these drugs have a narrow therapeutic index and the species and individual dose responses vary widely, clinicians are advised to start at the lower end of the dosage range. The intravenous route is preferred, as the dose can be titrated to effect.<sup>12</sup> Both xylazine and diazepam can be mixed in the same syringe with ketamine. Care must be taken, especially with smaller doses, to eliminate all air pockets from the syringe and to thoroughly mix the two drugs. Recovery from intravenous administration of these injectable anesthetic combinations may take 15-45 minutes, while recovery from intramuscular administration, especially if additional dosages have been necessary, may take hours. Recovery may be violent. Yohimbine has been shown to be an effective reversal agent for ketamine/xylazine anesthesia in raptors. A dosage of 0.1 mg/kg yohimbine was effective in reversing anesthesia caused by the administration of intravenous ketamine (4.4 mg/kg)/xylazine (2.2 mg/kg).<sup>5</sup> Tolazoline (15 mg/kg IV) has been

used to successfully reverse ketamine/xylazine anesthesia in turkey vultures.<sup>2</sup> The use of these reversal agents may make the use of ketamine/xylazine safer and more practical for birds.

Etorphine has been successfully used in large birds such as ostriches and cassowaries. A dosage of 0.02 to 0.03 mg/kg IM is utilized for restraint. This can then be reversed with 0.04 to 0.06 mg/kg IV of diprenorphine.<sup>15</sup>

Midazolam (same group as diazepam) (15 mg/kg IM) was successfully used to reduce the percentage of isoflurane necessary for general anesthesia in racing pigeons. It was then effectively reversed with the drug flumazenil (0.1 mg/kg IM).<sup>29</sup>

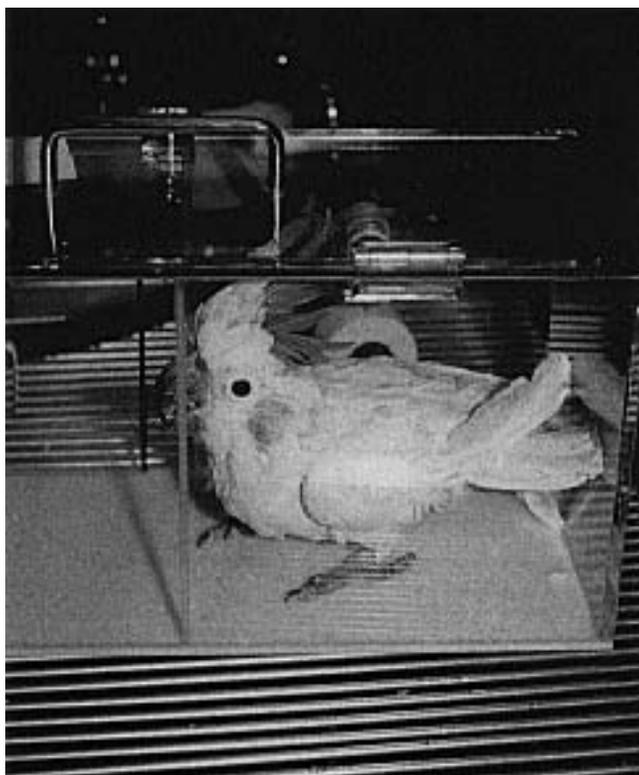
## Gas Anesthetic Equipment and Delivery

### Anesthetic Machines and Vaporizers

Most of the available models of anesthetic machines are adequate for an avian practice. What is required is an out-of-circuit, precision vaporizer for the administration of isoflurane. The vapor pressure of isoflurane (261 mm Hg) is so close to halothane (243 mm Hg) that the same type precision vaporizer can be used for both agents (Figure 39.2). However, once converted to isoflurane, a machine should no longer be used for halothane. A vaporizer can be purchased that is manufactured specifically for use with isoflurane, or a halothane vaporizer can be cleaned with ether and re-calibrated for use with isoflurane. A vaporizer cannot be used for halothane and isoflurane at the same time. Switching back and forth



**FIG 39.2** The vapor pressure of isoflurane and halothane are similar so isoflurane can be delivered through a halothane vaporizer; however, once a vaporizer has been converted to isoflurane use, it should not be used with halothane.



**FIG 39.3** Tank systems should not be used for inducing anesthesia in birds.

between these agents will destroy the vaporizer. Calibration of the vaporizer is necessary after a conversion has occurred from halothane to isoflurane. Once isoflurane is in use, the vaporizer should be cleaned and re-calibrated on a yearly basis.

### Breathing Systems

Gas anesthesia can be achieved through either an open, semi-open or semi-closed system. Open systems rely on the animal's being placed in contact with an absorbent material soaked in the anesthetic liquid. There are descriptions of methoxyflurane being administered in a drip cone system. With the highly volatile anesthetics like halothane and isoflurane, very high concentrations of the gas will rapidly occur in the inspired air, causing acute anesthetic overdose and death. Tank systems used to induce anesthesia in small mammals should not be used in birds. These chambers prevent monitoring of the patient, create a potential for beak, head, neck or spinal trauma and release high concentrations of gas into the environment when the top is opened (Figure 39.3).

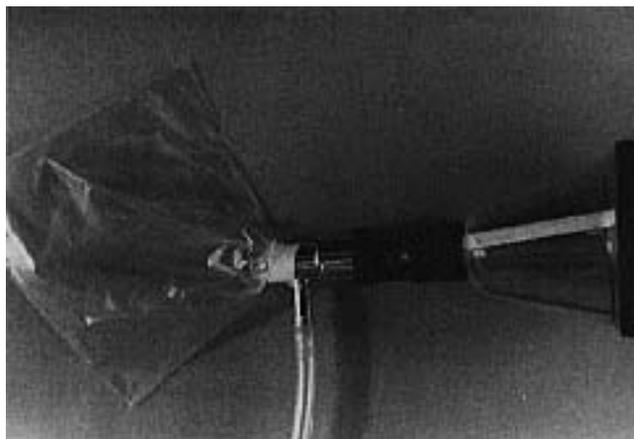
Semi-open systems rely on an Ayres T-piece, Y-piece, Norman elbow or Kuhn circuit that prevents the rebreathing of expired gases. Because of the rela-

tively low resistance to air flow present in these systems, they are ideal for avian patients.

Semi-closed systems rely on the complete rebreathing of expired gases. The resistance inherent in the circuit makes them impractical for birds.

A non-rebreathing anesthetic system is recommended for patients under seven to eight kilograms (most birds). This reduces dead space and decreases the effort that the patient must exert in order to breathe. This is especially important in birds, because both expiration and inspiration involve active use of the trunk muscles. Either an Ayer's T-piece or Bain's circuit can be effectively used with most birds. Some clinicians prefer the Bain's circuit because in theory, the patient's expired gases warm the in-flowing gases and reduce the loss of body heat. This can be critical in birds because their small size predisposes them to hypothermia, and respiration is one of the major routes through which body heat is lost. In patients over seven to eight kilograms, conventional human pediatric supplies are adaptable, easy to obtain and easy to maintain. In larger avian patients (eg, ostriches), standard small animal anesthetic equipment and supplies are applicable.

A standard 0.5 liter reservoir bag can be used in some larger avian patients, but better control and monitoring of respiration can be achieved with a smaller volume bag specifically designed for birds. These can be handmade from plastic bags, or an inexpensive, disposable avian anesthesia bag is commercially available (Figure 39.4).<sup>c</sup>



**FIG 39.4** Anesthesia bags (50 ml, 100 ml and 250 ml) are commercially available for use in birds. These inexpensive bags account for the reduced tidal volumes of birds, allow for precise IPPV and are disposable, which prevents nosocomial infections. The bags can be adapted to any Ayres T-piece-type semi-open anesthetic delivery system (courtesy of Exotic Animal Medical Products).

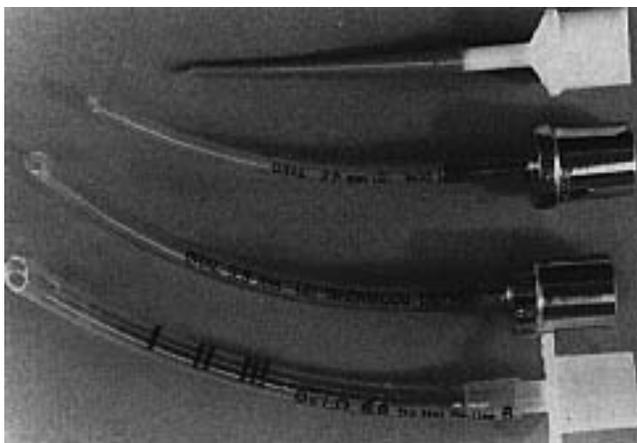
An appropriate scavenging system is the best protection for operating room personnel from secondary gas exposure. Once the scavenging system is in place, gas exposure can be reduced by quickly intubating patients, minimizing the time the patient is wearing a mask and keeping flow rates as low as possible to prevent gas from escaping via the pop-off valve. End valves placed in the reservoir bag can be vented directly into the scavenging system.

### Endotracheal Tubes

Non-cuffed infant, Magill or Cole (smallest size = 2 mm) endotracheal tubes can be used in medium- to large-sized birds (Figure 39.5). Cuffless tubes are used because birds have complete tracheal rings that cannot expand if excessive amounts of air are introduced into a cuffed tube. Alternatively, some clinicians choose to make their own endotracheal tubes out of red rubber feeding tubes. The end of the tube is snipped off and small holes are cut in the surface of the tube to allow for air exchange. The tip of the tube should be blunted by heating it with a flame and pressing it on a hard surface. These tubes are less costly than purchased tubes and have the added bonus of being disposable. In any situation, a tube with the maximum internal diameter that will fit in a bird's trachea should be used.

### Face Masks

The delivery of inhalant gases from a precision vaporizer can best be achieved by manually restraining the patient and placing the nostrils and mouth in a face mask connected to an Ayres T-piece anesthetic circuit. Common canine or feline anesthetic masks, while not ideal, can be used for induction. Most small



**FIG 39.5** Non-cuffed, 2 mm endotracheal tubes are commercially available. These are generally small enough for use in birds up to 150 g. In smaller birds, a red rubber feeding catheter with several holes cut in the end can be used as an endotracheal tube.

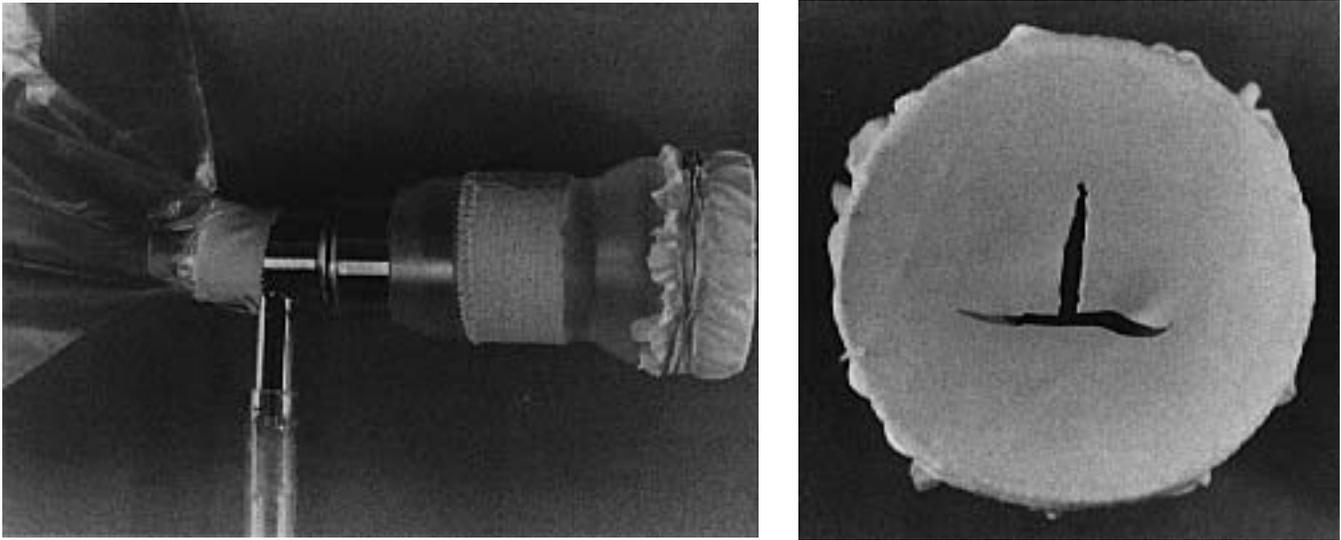
animal masks fit avian patients poorly, resulting in dilution of the anesthetic gas with room air. With these leaks, higher gas and oxygen settings are necessary in order to compensate for leakage (Figure 39.6). To avoid nosocomial infections, a disposable plastic drinking cup, with soft paper products placed between the cup and the patient's neck to prevent gas leaks, can be used as a face mask (Figure 39.7). In birds less than 150 g, an effective mask can be made by covering the end of a 12 cc syringe case with a section of latex glove. The syringe case can then be slipped over an Ayres T-piece with a 50 ml anesthesia nonrebreathing bag (Figure 39.8).



**FIG 39.6** A small animal face mask can be used to induce gas anesthesia in medium and large companion birds.



**FIG 39.7** Using a disposable plastic cup as an induction mask prevents the transmission of respiratory pathogens (eg, chlamydial, viral, fungal) between patients. If small animal face masks are used, they must be cleaned and sterilized between avian patients.



**FIG 39.8** A 12 cc syringe case can be cut to fit over an Ayres T-piece and covered with a latex glove to be used as a disposable face mask in small avian patients.

### Care of Equipment

The proper use and maintenance of anesthetic equipment is an often overlooked area.

With the large number of infectious bacterial, fungal and viral agents encountered in avian patients, any equipment used during anesthesia, including tubing and endotracheal tubes, should be thoroughly disinfected to reduce the chance of nosocomial infections. Equipment should not be used for other companion animals and then used for birds without sterilization. While the face mask and Ayres T-piece can be easily disinfected in cold sterilization solutions, anesthetic bags are much more difficult to disinfect.

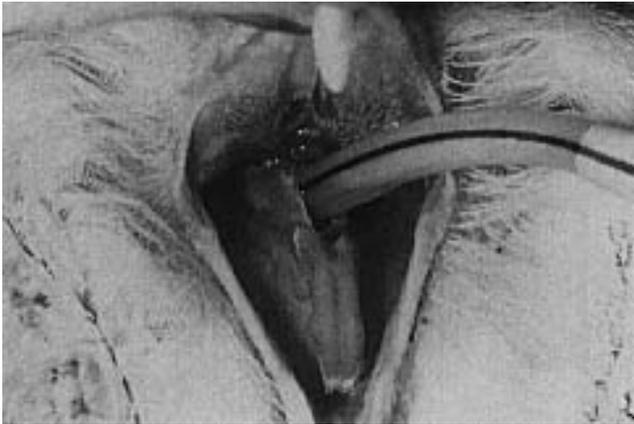
Tubing, reservoir bags, face masks and endotracheal tubes should be thoroughly cleaned with soap and water, then rinsed with clear water. They should then be disinfected using a chemical disinfectant and rinsed again with clear water. Finally, they should be allowed to air-dry in a clean, dust-free location. Alternatively, they may be sterilized using ethylene oxide or, with some endotracheal tubes, a heat autoclave. Because this cleaning regime must be used with every anesthetic episode, a large reserve of equipment is necessary to handle a sizable avian patient case load. Many clinicians feel it is more economical to use disposable anesthetic supplies than to use technician time for cleaning equipment. Disposables, however, are more expensive and they contribute to the medical waste problem.

### Delivery of Inhalant Anesthetics

Two methods of anesthetic induction with isoflurane have been discussed. One method is to place the bird in a face mask and slowly increase the gas to a level of 2.5 to 3%. However, the editors believe that the rapid induction achieved by using a 5% setting initially, followed by a decrease to maintenance levels of 1 to 2% is a better method. The amount of isoflurane delivered will vary with the patient, the individual anesthetic machine and the delivery system. Some macaws, owls and Galliformes appear to be particularly sensitive to gas anesthesia and may become apneic even with the use of isoflurane. Maintenance levels of anesthesia may be as low as 0.25% in sensitive individuals.

After induction, any patient that will be anesthetized for more than ten minutes should be intubated with an appropriately sized endotracheal tube (Figure 39.9). The amount of dead space should be minimized by ensuring an adequate gas flow and by using tracheal tubes of the proper length. The appropriate endotracheal tube length can be determined by measuring the distance from the thoracic inlet to the tip of the beak. The laryngeal structure of birds is highly mobile and can be manipulated from below the mandible to improve access for intubation.

Following intubation, the endotracheal tube can be connected directly to the semi-open system. If there is a possibility of regurgitation, the tongue and glottis should be pulled cranially, and the esophagus



**FIG 39.9** In some birds (like this owl), it is easy to visualize the glottis for intubation. In others (many psittacine birds), it is more difficult to visualize the glottis. Intubation can be simplified by placing a finger in the intermandibular space and gently lifting the glottis into view.

should be packed with moistened cotton to prevent aspiration.

#### Air Sac Administration

For surgery of the head, trachea or syrinx, anesthesia can be delivered by placing a short endotracheal or red rubber tube into the clavicular or caudal thoracic air sacs.<sup>25,26</sup> The placement procedure is rapid and may be indicated as an emergency tactic when an animal is presented with respiratory arrest or when severe dyspnea has been induced by foreign body aspiration. To place an air sac tube, the animal is positioned with the leg extended to the rear as for a surgical sexing procedure. A small skin incision is made over the sternal notch, and hemostats are used to produce an entrance through the body wall and into the left abdominal air sac (see Chapter 13). A shortened endotracheal tube can then be inserted into the air sacs and the Ayres T-piece connected directly to the tracheal tube.

A study in pigeons and common buzzards indicated that isoflurane could be safely administered through the air sacs to maintain a surgical plane of anesthesia for 60 minutes. These birds were monitored for changes in temperature, pulse rate, oxygen saturation, blood pressure and  $\text{paCO}_2$  levels (Table 39.1). In this study a persistent period of apnea was noted when the air sac tube was perfused with oxygen. It was suggested that the loss of  $\text{CO}_2$  present in the lungs and air sacs decreased the stimuli to the respiratory centers and induced the apnea. No assisted respiration techniques were used during the 60-minute period of apnea.<sup>16</sup>

At the end of the 60-minute study, the birds were flushed with oxygen to remove residual isoflurane and their  $\text{paCO}_2$  levels dropped to 19. This hypocapnia was followed in four minutes by the return of spontaneous respiration, which was associated with an increase of  $\text{paCO}_2$  levels (40 in the pigeons, 31.7 in the buzzards). Arrhythmias were common in the pigeons starting around 50 minutes after induction. The part that hypothermia played in inducing arrhythmias could not be determined. The mean recovery time to standing was 10 to 12 minutes. A metabolic alkalosis occurred throughout the anesthetic episode that was attributed to the decrease in  $\text{paCO}_2$ .<sup>16</sup>

**TABLE 39.1** Mean Effects of Air Sac Anesthesia with Isoflurane<sup>16</sup>

	Pigeon	Buzzard
Temperature (°C)	37.3 (41.5)	38.7 (41)
ApH	7.62 (7.36)	7.67 (7.51)
$\text{paO}_2$	426 mm Hg	360 mm Hg
$\text{paCO}_2$	19.2	19.1
Pulse rate	93.1 (172)	232 (301)
Blood pressure	64.6 mm Hg (106)	119.7 mm Hg
$\text{O}_2$ saturation (pulse oximeter)	98.1%	99%

Normal is listed in parenthesis. ApH = arterial pH,  $\text{paO}_2$  = partial pressure of oxygen,  $\text{paCO}_2$  = partial pressure of carbon dioxide.

## Management of the Anesthetic Patient

### ■ Patient Evaluation

Isoflurane can be used for two distinct purposes in the avian practice. One is for long-term general anesthesia needed to perform difficult diagnostic procedures and surgeries. The other is as an anesthetic to provide short-term restraint and to facilitate the ease of performing fluid administration, blood collection, radiography, endoscopy or bandage changes (Figure 39.10).

The goal of a preanesthetic evaluation is to detect and correct any underlying pathology prior to inducing anesthesia. In some cases, several days or weeks of supportive care may be needed in order to ade-



**FIG 39.10** Isoflurane is generally safe to use for short procedures (eg, radiography, endoscopy, blood collection) in patients with only a history and a physical examination. If a bird will be anesthetized for more than ten minutes, a complete database should be obtained to assess the stability of the patient.

quately stabilize a compromised patient before anesthesia can be safely administered. In other cases, eliminating abnormalities may not be possible, and consequently, adjustments must be made in inducing and monitoring anesthesia to ensure a successful outcome.

The minimum database is designed to help the anesthetist develop a list of risk factors associated with general anesthesia in a given patient and to develop evaluation methods or supportive care techniques that decrease the anesthetic risk. The patient evaluation associated with the use of isoflurane for short-duration procedures (five to ten minutes) is generally limited to a thorough history and physical examination. The theory in using isoflurane for short-term procedures with a limited database is that this drug has a wide therapeutic index and has consistently been shown to be relatively safe even in severely compromised patients.

If a bird is going to undergo anesthesia for more than ten minutes, then in addition to the history and physical examination, a minimum database ideally should include a fecal parasite check, Gram's stain of feces and choana, PCV, TP, WBC with differential, a platelet estimate, estimated clotting time, bile acids, AST, LDH and UA. Additional tests may be indicated depending on the initial assessment and the type of procedure requiring anesthesia.

### Risk Classification

Successful risk classification depends on conservative evaluation of the minimum database. Practically, risk assessment allows one to estimate the duration of safe anesthesia, the intensity of monitoring required and the supportive care and technical assistance needed to ensure patient safety. It also provides the clinician with a prognostic perspective to be discussed with the owner (see Table 39.2).

It can be successfully argued that few avian patients requiring anesthesia are Class I. Most patients are Class IV and V. With careful supportive care, a clinician can reduce the anesthetic risk factors of many Class IV and V patients to those of Class II or III patients.

A commonly encountered complication of anesthesia in birds is liver dysfunction. Hepatopathies may be suspected when performing the initial physical examination and confirmed with findings of lowered protein levels, elevated or depressed bile acids and tissue enzyme values, and radiographic findings of hepatomegaly, or conversely, a loss of liver mass. Liver dysfunction may reduce a patient's ability to metabolize anesthetic agents and may also be associated with coagulopathy. The use in these patients of any anesthetic drug that is dependent on the liver for metabolism is contraindicated (most injectables, halothane, methoxyflurane). Supportive care designed to improve liver function may be necessary before anesthesia. If anesthesia cannot be postponed to allow

**TABLE 39.2 Risk Classification of Potential Anesthesia Patients**

<b>Class I</b> (minimal risk)	Young, healthy patient undergoing an elective procedure.
<b>Class II</b> (some risk)	Young, healthy patient undergoing a non-elective procedure, or a healthy patient undergoing an elective procedure.
<b>Class III</b> (risky)	Patient with an ongoing health problem undergoing a procedure for this or another problem.
<b>Class IV</b> (very risky)	Patient with a major health problem (unstable in nature) undergoing a procedure.
<b>Class V</b> (moribund)	Last ditch effort to save bird's life.

sufficient stabilization, then anesthetic agents that are minimally metabolized by the liver (eg, isoflurane) and vitamin K injections to help promote clotting are suggested.

Obesity is common in companion birds. Excess fat deposits interfere with the patient's ability to ventilate. Respiratory effort is further compromised when a bird is anesthetized and placed in abnormal body positions for surgery. With elective procedures, dietary changes and treatment for underlying metabolic disorders are indicated before general anesthesia is performed.

Surgery is frequently required for egg-related peritonitis, especially in small species (eg, cockatiels, lovebirds, budgerigars). These patients should be carefully stabilized prior to anesthesia. This stabilization process may take weeks and in some cases render surgical intervention unnecessary. An abdominal tap and diuretic therapy are indicated prior to anesthesia to reduce the volume of fluid in the abdomen and to improve the patient's ability to breathe.

These patients should be maintained in an upright position to prevent abdominal fluid and debris from entering the lungs through rents in the air sacs. The surgery table should be tilted so that the patient's head is slanted up.

In non-elective procedures on obese patients or those with an abdominal fluid accumulation, proper intubation is mandatory to ensure a patent airway. Gentle IPPV will help maintain adequate oxygenation. Keep in mind when providing IPPV that the avian lungs inflate minimally, and pressure placed on a breathing bag should be less than 15 mm H<sub>2</sub>O to prevent rupture of the air capillaries.

## Preanesthetic Stabilization and Preparation

### Nutritional Therapy

For elective procedures, inadequate diets should be corrected three to four weeks before surgery. For specific nutritional requirements and nutritional support of surgical patients, see Chapters 3, 15 and 40.

### Fluid Therapy

Fluid therapy is an important aspect of supportive care that should be provided to avian patients. Many sick birds presented for evaluation have been off food and water for at least a day, often longer. With their high metabolic rate, birds rapidly become dehydrated. Correcting dehydration will dramatically im-

prove the patient's ability to physiologically cope with anesthesia.

It has been suggested that all birds suffering from trauma and disease can be assumed to be at least ten percent dehydrated, and that the following formula should be used to calculate their fluid requirements: normal body weight (grams) x 0.1 (10%) = fluid deficit in ml. The dose for maintenance fluid is estimated at 50 ml/kg/day.<sup>24</sup>

Calculated fluid requirements can be administered in severely dehydrated patients by an intravenous or intraosseous route (see Chapter 15) (Figure 39.11).<sup>17,30</sup> Fluids administered by oral or subcutaneous routes are not as effective in restoring or maintaining circulating volume.



**FIG 39.11** Placing an intraosseous cannula in the tibiotarsal bone is an excellent way to provide fluids to a patient during surgery. In comparison to catheters in peripheral vessels, intraosseous cannulas are much easier to maintain. Intraosseous cannulas placed in the tibiotarsus can also be used in critically ill patients for intermittent fluid, blood or drug administration.

Lactated Ringer's solution is the fluid of choice. This balanced electrolyte solution protects renal function better than sugar solutions.<sup>23</sup> Five percent dextrose is not a satisfactory replacement solution because the dextrose is metabolized, leaving free water.<sup>23</sup> Fluids administered to anesthetized birds should be heated (96°F) to prevent hypothermia.

Fluids must be carefully administered to birds to prevent volume overload. The maximum acute fluid load that can be tolerated by healthy patients is 90 ml/kg/hr.<sup>14</sup> In a cockatiel this would be a maximum of 9 mls in an hour or 0.15 mls per minute. Most avian patients, however, are unstable and cannot tolerate such a high rate and volume of fluid administration.

Electrolyte levels and acid-base balance should always be of concern in the anesthetic candidate. Some newer analyzers use small volumes (10 µl) of serum, providing clinicians access to actual values to use in assessing the status of their patients. If no means of determining the bicarbonate deficit is available and the patient is dehydrated or critically ill, the administration of 1 mEq/kg of bicarbonate at 15 to 30 minute intervals to a maximum of 4 mEq is suggested.<sup>24</sup>

### Fasting

Recommendations concerning fasting of birds prior to anesthesia have varied from no fasting to an overnight fast.<sup>12,13</sup> Practically speaking, the patient should be kept off food long enough for the upper gastrointestinal system to become empty. This process takes overnight in large birds and four to six hours in smaller birds. One should palpate the crop and postpone surgery if it is not empty. In an emergency, a patient with food in the crop should be held upright during the induction procedure, with a finger blocking the esophagus just below the mandible. Once the animal is anesthetized, the crop can be emptied by placing a finger covered with cotton or gauze over the choanal slit to prevent food entering the nasal cavity, turning the bird upside down and manually emptying the crop and esophagus. The esophagus can then be packed with gauze, and the head and neck positioned on an upward slant to minimize the chances of passive regurgitation. The trachea should be intubated using an appropriately sized tube.

### Patient Monitoring

The goal in any anesthetic episode is to maintain the lowest possible level of anesthesia to achieve necessary restraint. Anesthetic planes in birds are difficult

to observe from outward signs, and depth should be monitored by combining information obtained from heart rate monitored by either an ECG or doppler, respiratory rate and effort, toe pinch, palpebral and corneal reflexes and wing tone. The rates will vary depending on the species (Table 39.3). A doppler can be placed on either the cranial tibial or medial metatarsal arteries.

**TABLE 39.3 Heart Rates and Respiratory Rates in Birds Anesthetized with Isoflurane**

	Beats Per Minute	Breaths per Minute
Budgerigar	600-750	55-75
Cockatiel	450-604	30-40
Pigeon	93.1±5.4 <sup>16</sup>	15-25
Parrot	120-780	10-20
Ostriches <sup>4</sup>	60-72	2-20

Anatomic locations to evaluate the reflex response to pain or touch in avian patients include palpebra, cornea, cloaca, propatagium, cere, interphalangeal area, pupils (response to light) and pectoral muscles.<sup>16</sup>

In a light plane of anesthesia, the patient has a palpebral, corneal and pedal reflex but has lost voluntary motion. The ideal anesthetic level, as described in one study,<sup>16</sup> was when the bird's eyelids were completely closed and mydriatic, the pupillary light reflex (pupillary response to light) was delayed and the nictitating membrane moved slowly over the entire cornea. The muscles were relaxed and all pain reflexes were absent. The loss of a corneal reflex (no reflex closure of the lid after touching the peripheral cornea with a dry swab) was considered to indicate deep anesthesia.<sup>16</sup>

The respiratory rate should be slow and deep. If a patient becomes too deep, all reflexes will be lost and the respiratory rate will be slow and irregular. Wing flutter is often an early indicator that an animal is

### CLINICAL APPLICATIONS

#### Anesthetic planes may be monitored by:

- Rate and depth of respiration
- ECG
- Heart rate — doppler
- Toe pinch
- Palpebral reflex
- Temperature
- Wing flutter
- Lid closure
- Pupillary dilation
- Pupillary reflex
- Corneal reflex

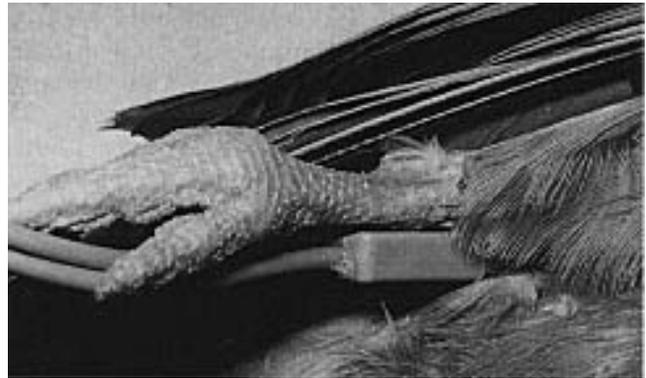
becoming light. An excellent plane of anesthesia for most procedures can be accomplished by reaching a depth of anesthesia where wing tone has just disappeared. If injectable agents have been used, the traditional planes of anesthesia may not be present, making evaluation of the patient more challenging.

### Body Temperature

Physiologically, birds are actually less efficient homotherms than mammals and as a result undergo more rapid changes in body temperature during anesthesia. Loss of heat during surgical anesthesia is a very important factor in anesthetic survival and in the rate of return to a physiologic normal state following anesthesia. Besides a loss of physiologic responses to reduced core temperature, hypothermia is also induced during surgery by removal of large areas of feathers to expose surgical sites, by the constant flow of cool anesthetic gases through the respiratory tract, by the liberal use of alcohol, by body contact with a cold conductive surface and by the length of the procedure. Even with supplemental heat, it is not unusual to have rapid reductions in core temperature during anesthesia. Any degree of hypothermia compromises the patient's ability to recover from anesthesia. While supplemental heat will not prevent the drop in core temperature seen with anesthesia, it does tend to reduce the speed of heat loss.

All patients undergoing long surgical procedures should be placed on water circulating heating pads. Keep in mind that these devices need at least a 20-minute warmup period before they reach the preset temperature. The clinician may choose to minimize the amount of alcohol used in the surgical scrub and instead use chlorhexidine or povidone iodine to minimize heat loss through evaporation. Body temperature loss may also be minimized through the use of heat lamps, heated lavage solutions, heated IV fluids or hot towels wrapped around non-rebreathing tubes. Hypothermia occurs in birds despite their being placed on water circulating pads.

Body temperature (normal=105 to 107°F<sup>28</sup>) can be constantly monitored during anesthesia to properly evaluate the degree of heat loss. A patient under anesthesia will experience a time-related reduction in body temperature (maybe as much as 10°F), which can predispose to cardiac arrhythmias and increased recovery times. If severe hypothermia (loss of greater than 10°F) occurs, the patient may not recover at all. Traditional thermometers can be used to record this data, but they require cloacal insertion and take



**FIG 39.12** A doppler probe placed on the medial metatarsal artery can be used to monitor the heart rate during anesthesia.

three to five minutes to give an accurate reading. Electronic thermometers also require cloacal insertion and take two to three minutes to give a reading. A more practical option is a tympanic scanner,<sup>d</sup> which gives a reading in five to six seconds. The monitors are easy to use by applying the probe to the outer surface of the ear. Tympanic temperatures are consistently slightly higher than cloacal temperatures.<sup>21</sup>

### Heart Monitoring

Heart rate in avian patients can be monitored in several ways. Palpation at the point of maximum intensity is possible in some patients (see Table 39.3). Peripheral pulses can be used but are often difficult to detect in smaller patients. Auscultation via a stethoscope is possible, although the chest may be difficult to reach once the patient is draped for surgery. Esophageal stethoscopes equipped with pediatric tubing can be used for auscultation of the heart in avian patients the size of Amazon parrots and larger. This monitoring system allows direct auscultation of the heart without having to go near the surgical field. If correctly positioned, the esophageal stethoscope can also be used for monitoring respiration. Dopplers can be used, but are extremely positional in nature and difficult to maintain in birds (Figure 39.12). Some oximetry units provide pulse rates up to 250 bpm; these units are easy to use and are not positional like the doppler. In a group of cockatiels maintained in a surgical plane of anesthesia, the heart rate remained above 450 bpm.<sup>12</sup> In one study with pigeons and common buzzards, the pulse rates determined by the oximeter were consistent with those determined through a direct arterial line.<sup>16</sup> In an emergency situation, a heart rate can be quickly obtained by placing the doppler probe on the globe of the eye. An ECG is an excellent way to monitor the

depth of avian anesthesia. As a bird gets deeper, the T-waves become smaller and may totally disappear. As the depth further increases, the R-wave will increase in magnitude and the S-wave is reduced.

### Respiratory Rate

Respiratory rates during anesthesia should be slow and regular (see Table 39.3). Hyperventilation or rapid jerky motions are indications of ensuing problems. An increase in the respiratory rate may indicate that a bird is entering a lighter plane of anesthesia, that the bird is having difficulty breathing (occluded tracheal tube) or that the bird has an elevated  $\text{paCO}_2$ . Because surgical patients are draped, it is often difficult to observe respiratory effort. Direct visualization of chest movement as an indication of respiration can be facilitated by using clear sterile surgical drapes.<sup>e</sup> An anesthetic bag<sup>e</sup> designed to deal with the lower tidal volumes of birds will also make it easier to monitor respiration.

When using halothane or methoxyflurane, apnea and cardiac arrest may develop at the same time without prior warning. With isoflurane, apnea usually proceeds cardiac arrest by several minutes. Halothane typically induces a rapid decrease in heart rate that returns to normal shortly after ceasing gas administration.

Apnea monitors do work in birds; however, less expensive units may not be sensitive enough to detect the respirations of smaller patients. The monitoring device that attaches to the endotracheal tube is heavy, and precautions must be taken to keep the weight of the monitor from kinking small diameter tubes. These units also require that the patient be intubated. A unit intended for use in birds should be evaluated on avian patients prior to purchase.

### Blood Pressure

Blood pressure can be monitored directly or indirectly and should remain above 100 mm Hg. Except in a research situation, it is unlikely that most practitioners are going to opt for direct blood pressure monitoring due to the cost and invasiveness of the procedure. Although indirect monitors that use pediatric cuffs are available and offer some promise for

use in avian patients, they are less reliable for smaller patients. Appropriate responses to patients with decreased blood pressure values include reduction of anesthetic levels, administration of IV fluids, correction of hypothermia and evaluation and correction of blood loss.

### Blood Gases

Arterial blood gases can be assessed directly by running arterial samples on a blood gas machine. This is not practical for most practitioners and it may involve taking multiple blood samples, a procedure that smaller patients cannot tolerate. Indirect assessment of blood oxygenation can be done via an oximeter. An oximeter records the percentage of circulating oxygenated blood via a noninvasive probe.<sup>10</sup> The unit works by spectrometrically measuring the difference in absorbance between reduced hemoglobin and oxyhemoglobin. Oximeters are not as sensitive at direct determination of arterial blood gases, and oximeter readings at 98% saturation are common in birds that are actually 100% saturated.<sup>16</sup> The author has found that the oximeter probe can be attached to the wing web, toe, tongue or the area over the tibiotarsal bone. The tibiotarsal area seems to give the most consistent and reliable readings. The probe is small and requires only one wire so it is easily used during surgery. Additionally, many oximeters are actually pulse oximeters that produce an audible beep, allowing the anesthetist to monitor heart rate as well as oxygen saturation levels. Ideally, oxygen saturation levels should be above 90%. A reading below 80% should be considered life-threatening. Most birds will maintain an oxygen saturation between 80 to 85% when self-ventilating. Use of IPPV to increase oxygen saturation is, therefore, valuable. Normal blood gas values of  $\text{paCO}_2=19$ ,  $\text{paO}_2=400$  and  $\text{ApH}=7.4$  have been reported.<sup>20</sup>

### Anesthetic Emergencies

With careful assessment of patients, conscientious supportive care and thorough monitoring, many anesthetic-related emergencies can be prevented.

### Respiratory Arrest

If respiratory arrest occurs, the anesthetic system should be disconnected from the bird and the chest should be lightly pressed and released to induce air intake, or fresh air can be gently delivered into the tracheal tube. If the patient is not intubated, an air sac tube should be placed or the animal should be immediately intubated. The practitioner must keep mind that air sac intubation may result in apnea while the  $\text{paO}_2$  is increasing and the  $\text{paCO}_2$  is decreasing.

## CLINICAL APPLICATIONS

### Emergency Steps for Respiratory Arrest

- Disconnect bird from anesthetic
- Press on sternum 40 to 50 cycles/min
- Intubate or apply air sac tube
- IPPV through tube

If respiratory arrest occurs in response to injectable anesthetics, the reversal agent should be administered intravenously. The administration of doxapram HCl on the tongue may help stimulate respiration. The pulse rate should be carefully monitored and resuscitation should continue until the bird is breathing unassisted. Birds that show respiratory arrest should be rescheduled for the procedure; a second or third episode of apnea in these cases is often followed by cardiac arrest.

### Cardiac Arrest

Cardiac arrest represents a poor prognosis. Resuscitation efforts are often unsuccessful. If success is to be achieved and cardiac arrest truly does exist, the clinician should be aggressive and utilize open heart massage. The rate should be 60 or more compressions per minute and they should be accompanied by coordinated artificial ventilation. These efforts should be continued for up to five minutes. Although rare, some birds may recover following cardiac arrest.

### Hemorrhage

If hemorrhage occurs during surgery, a significant portion of that loss can be replaced via fluid therapy using isotonic solutions. If hemorrhage is severe, a transfusion will be necessary (see Chapter 15).

### Postanesthetic Monitoring and Recovery

Anesthetic recovery should occur in a pre-heated environment, preferably a pediatric or avian incubator. The

safest approach to post-anesthetic monitoring of a patient is to leave all of the monitoring devices in place until the patient absolutely will not tolerate them. Patients should be recovered where they can be easily observed. Birds should not be left unsupervised until they are perching without difficulty.

Recovery from injectable anesthetics will be much more prolonged and if ketamine has been used, potentially traumatic. Anesthetic recovery is best accomplished by wrapping the bird in a towel to prevent wing-flapping and self-inflicted trauma. The lights should be dimmed and the noise level kept to a minimum to prevent violent reactions. The patient should be rolled over every few minutes, and the pharynx should be monitored for the accumulation of mucus or vomit. In severely depressed birds, IPPV can be continued until the patient is no longer willing to tolerate the tracheal tube. Recovery from even long anesthetic episodes with isoflurane generally requires less than five minutes. The administration of a reversal agent is indicated when appropriate.

#### Products Mentioned in the Text

- a. AErrane, Anaquest, Madison, WI
- b. IsoFlo, Solvay Animal Health, Mendota Heights, MN
- c. Avithesia, Exotic Animal Medical Products, Watkinsville, GA
- d. Tympanic Scanner, Exergin Corp, Newnan, MA
- e. Steri-drapes, 3M, St. Paul, MN

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