ARTERIAL CATHETERIZATION, INTERPRETATION, AND TREATMENT OF ARTERIAL BLOOD PRESSURES AND BLOOD GASES IN BIRDS

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Abstract

Blood pressure monitoring of patients has become increasingly common in companion animal veterinary hospitals, especially during anesthesia, surgical procedures, critical care, and general health assessments. Determining an animal's blood pressure has become a standard part of the routine diagnostic evaluation for monitoring hypertension in geriatric patients or patients affected with renal insufficiency, cardiac disease, vision loss, or endocrine disorders. To increase the standard of care in exotic and zoological medicine, new diagnostic techniques must be identified and implemented. Blood pressure measurements in avian species are more challenging because only direct blood pressure techniques have been reported to be accurate. Arterial catheterization and interpretation can be daunting without the knowledge of avian physiology and anatomy; however, techniques for placing arterial catheters are not difficult once clinicians have gained sufficient experience. This article describes the techniques, anatomy, and appropriate interpretation of blood pressure results obtained through arterial catheterization in birds.

Key words: anesthesia; arterial catheterization; avian; blood pressure; monitoring

Blood pressure assessment is a crucial component of patient care during anesthesia, surgery, critical care, and general health assessments. Determining a patient's blood pressure has become a standard procedure in companion animal medicine for monitoring hypertension in geriatric patients or animals affected with renal insufficiency, cardiac disease, vision loss, or endocrine disorders. In addition, monitoring a patient's blood pressure has become part of standard protocol during anesthetic procedures and ideally should be performed for every patient.[1]

Avian blood pressure monitoring is a useful diagnostic tool in the assessment and treatment of hypertensive and hypotensive diseases in psittacine patients. Multiple retrospective studies involving psittacine species have shown a high prevalence of atherosclerosis. One study in Amazon and African gray parrots described lesions in more than 91% of the birds evaluated.[2] Another study in psittacine birds showed a high prevalence of atherosclerosis that increased with age and female sex. In human medicine, studies have established a relationship between systemic hypertension and the development of atherosclerosis.[3,4] Detection of hypertension in animals could provide clinical

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insight into an animal's cardiovascular condition and establish risk factors and management protocols for atherosclerosis in birds.

Monitoring blood pressure has many advantages when treating critically ill psittacines as well as those that are high-risk candidates for general anesthesia. Accurate blood pressure monitoring may be extremely helpful in assessing the progress of fluid resuscitation and inotropic or pressor therapy in patients with hypovolemic or septic shock. Arterial catheters can be used for periodic assessment of blood gases, which can be especially beneficial in patients with respiratory disease or acid-base disorders.[5]

Currently, routine blood pressure monitoring is rarely used in avian patients owing to their unique physiology and anatomy and the difficulty of catheter placement. In veterinary medicine, there are typically 2 techniques available for measuring blood pressure: invasive and noninvasive methods. Invasive techniques require an arterial catheter. Noninvasive techniques include Doppler, photoplethysmographic/photoacoustic probes with a sphygmomanometer, and oscillometric monitors.[5]

In previous studies, noninvasive techniques have been reported to be inaccurate in measuring blood pressure in psittacines when compared with invasive blood pressure monitoring. A study performed on anesthetized Hispaniolan Amazon parrots, *Amazona ventralis*, showed low agreement between noninvasive blood pressure measurement using the pectoral limb and hindlimb when compared with direct arterial blood pressures.[6] Several factors can affect the accuracy of noninvasive blood pressure measurement in any species. These include the type of blood pressure monitor used, cuff size in relation to the limb being measured, site of cuff placement, and the patient's blood pressure.[7,8] The inconsistency of measurements between limbs observed in the aforementioned study may have been because of differences in avian anatomy. In the pectoral limb, birds have a propatagium (a web of skin), muscles, and tendons that connect the scapulohumeral joint to the radiocarpal joint of the pectoral limb. This protopatagium could potentially interfere with appropriate occlusion of the ulnar artery, leading to an inaccurate noninvasive blood pressure.[9] Moreover, birds normally have higher heart rates and blood pressures than most domestic animals, which may also influence the accuracy of noninvasive blood pressure results, in that indirect monitors may not be adequately calibrated to measure the higher values.[10]

Accurate determination of blood pressure is critical in any species. Invasive (direct) blood pressure measurement is considered the gold standard and has been shown to correlate well with systemic blood pressure.[11] Unfortunately, invasive blood pressure measurements can be adversely affected by numerous factors. The regional insertion of an arterial catheter affects blood pressure measurements. As a result of Ohm's law, the flow of blood between 2 points is equal to the pressure difference divided by the resistance offered by the blood vessel; therefore, blood pressure measurements are often higher in smaller peripheral arteries when compared with that of larger central vessels. Similarly, impedance, the pressure exerted by blood against the vascular walls, also increases with a reduction in vessel diameter. Thus, the farther an artery is from the aorta, the higher the recorded blood pressure. Compounding accurate measurement further, high resistance distal to the site of arterial pressure measurement may also lead to inaccurate readings owing to reflections of the pulse waves erroneously raising direct systolic pressures. Differences in compliance of the catheter wall, small iatrogenic air bubbles within the line or catheter, and partial occlusion of the system also cause false alterations in blood pressure measurements. The size of the catheter can also affect the accuracy, whereby inappropriate dampening of the arterial pulse signal can occur owing to the differences in the length and diameter of different arterial catheters.[12]

Avian physiology may also affect the interpretation of a patient's blood pressures. Avian vascular anatomy differs from that of mammals and reptiles. Bird arteries have a higher resilience, which may affect how quickly vessels respond to changes in cardiac output and resultant blood pressures.[13] A study in turkeys described arterial resilience between 85% and 87%, which was higher than values reported in most mammals.[14] In addition, unlike mammals, bird's arterial lamellar units in the tunica media do not form complete cylinders. Rather, their lamellae consist of a series of rigid and elastic components, which allows the arterial wall to have greater distention.[15] Owing to this increase in compliance, avian blood vessels often have much thicker walls when compared with mammalian vessels of the same diameter. Because of these adaptations, birds are inclined to have a lower total peripheral resistance and higher arterial pressure than mammals.[16] Birds frequently have larger hearts, higher stroke volumes, and lower heart rates than mammals of corresponding body mass.[14] These anatomic and physiologic differences ultimately allow sufficient cardiac output to meet their greater metabolic needs. Unfortunately, the importance of the physiologic differences between birds and mammals, as they relate to peripheral blood pressure determination and interpretation, is unclear.

Multiple studies on avian arterial blood pressure measurements have demonstrated that arterial blood pressure in most avian species is significantly higher when compared with that of mammals.[1,9,17-22] However, there have only been a few studies in birds that have determined the correlation between measured blood pressure values and the inhibition and autoregulation of the vascular system. A study performed on anesthetized galliformes comparing glomerular filtration rate and blood pressure found that galliformes were able to maintain their glomerular filtration rate when mean arterial pressure (MAP) ranged between 60 and 110 mm Hg.[23] When MAP decreased to less than 50 mm Hg, chickens were unable to sustain glomerular filtration and urine output ceased.[24] Unlike chickens that have normal systolic, mean, and diastolic arterial blood pressures of 99 ± 13, 84 ± 13, and 69 ± 15 mm Hg, respectively, values for normotension are higher in psittaciformes, gruiformes, falconiformes, accipitriformes, strigiformes, and other galliformes (e.g., turkeys).[10,20] In Hispaniolan Amazon parrots anesthetized with 2.5% isoflurane, the systolic, mean, and diastolic arterial blood pressures were 132.9 ± 22.1, 116.9 ± 20.5, and 101.9 ± 22.0 mm Hg, respectively.[25] Alterations in cardiovascular physiology between these avian species may contribute to the significant difference in blood pressure values. Additionally, most baseline avian blood pressure values are obtained from birds under anesthesia, where materials such as inhalant gases can cause significant depressant effects and can alter arterial blood pressure, either through reduction in systemic vascular resistance or a reduction in cardiac output.[26] However, when comparing anesthetized values to those of awake falconiformes, accipitriformes, and strigiformes, similar direct blood pressure values were obtained (Table 1).[10] Other than on galliformes, no studies have been performed describing the effects of different values of blood pressure on end-organ perfusion in avian species. Hypotension in humans has been described as a reduction of 30% from the baseline of conscious MAPs.[27] If this definition is used in a cross-species manner, the level of blood pressure at which birds are considered hypotensive would have a tendency to be higher than that recorded in mammals, with the exception of some galliforme and anseriforme species.

ARTERIAL CATHETER PLACEMENT

Unlike domestic mammals, there are only a few placement sites available for arterial catheterization in birds. For medium to large birds (>200 g), the deep radial artery is the preferred site. However, for smaller birds (<200 g), the authors favor catheterization of the superficial ulnar artery. For water birds or long-legged birds, the cranial tibial or dorsal metatarsal arteries are acceptable choices for catheterization. Catheterization of the external

TABLE 1. Evaluation of indirect blood pressure monitoring in awake and anesthetized red-tailed hawks (*Buteo jamaicensis*): effect of cuff size, cuff placement, and monitoring equipment.[10] Previous published direct blood pressure (DBP) values in avian species. Mean ± SD values for systolic arterial pressure (SAP), mean arterial pressure (MAP), and diastolic arterial pressure (DAP) are presented

Species	SAP ± SD	MAP ± SD	DAP ± SD	Experimental Methods	References
Chicken	99 ± 13	84 ± 13	69 ± 15	Sevoflurane	Naganobu et al[23]
Pigeon	93 ± 10	82 ± 14	72 ± 13	Isoflurane	Touzot-Jourde et al[28]
	88 ± 11	75 ± 10	60 ± 11	Isoflurane	Touzot-Jourde et al[28]
Sandhill crane		205 ± 29		1× MAC isoflurane	Ludders et al[21]
Cockatoo		143 ± 4		Isoflurane	Curro et al[29]
Amazon parrots	163 ± 18	155 ± 18	148 ± 18	2% Isoflurane	Acierno et al[9]
	132.9 ± 22	116.9 ± 20.5	101.9 ± 22.0	2.5% Isoflurane	Schnellbacher et al[25]
Great horned owl	232 ± 37	203 ± 28	178 ± 25	Baseline awake values	Hawkins et al[1]
Red-tailed hawk	220 ± 51	187 ± 42	160 ± 45	Baseline awake values	Hawkins et al[1]
Bald eagle	194 ± 13		158 ± 13	Isoflurane	Joyner et al[18]
	146 ± 13		135 ± 13	Sevoflurane	Joyner et al[18]

SD, standard deviation; MAC, minimum alveolar concentration.

carotid artery has also been described in birds, but it is usually more invasive and requires a cut down for proper visualization.[30] The clinical techniques of catheterization of the deep radial artery, superficial ulnar artery, and the cranial tibial or dorsal metatarsal arteries have been described in this article.

The authors prefer the deep radial artery to the other sites for arterial catheterization because of its minimal mobility. The deep radial artery lies just between the tendons of the extensor *digitorum longus* and the flexor *digitorum profundus* in the distal wing of most birds. Catheter placement should occur where the artery is most superficial at the ulnar carpal bone or at the distal head of the ulna. At this location, the deep radial artery can be readily found and palpated on the medial side of the wing. At a more proximal location, the artery dips deeper and runs along the radius.[31] Proximal placement of the catheter becomes more reliant on palpation skills of the veterinarian as the artery becomes less visible (Fig. 1). Moreover, with a more proximal placement, additional care is required during catheter insertion because of the close association of the deep radial artery with the median nerve.

The superficial ulnar artery, along with the recurrent ulnar artery, branches off from the ulnar artery at the elbow. The superficial ulnar artery then runs medially over the extensor *metacarpi radialis*, pronator *superficialis*, and pronator *profundus* muscles before dipping along the ulna and crossing the carpus and terminating at the base of the distal phalanx of the major digit (Fig. 1).[31] Although more prominent in size, the superficial ulnar artery is very mobile as it crosses the elbow, thus increasing the technical difficulty of placement without inducing hematoma. Furthermore, owing to the wing's unique anatomy, securing the catheter at this location can also be problematic.

The cranial tibial artery is the major vascular supply to the lower leg and its digits. As the artery dorsolaterally crosses the hock, it becomes the metatarsal artery, which then travels medially across the tarsometatarsus (Fig. 2).[31] The artery is more prominent and easier to visualize and palpate in long-legged birds. As the artery moves distally, catheterization becomes more problematic because of the keratinized scales on the bird's legs. As most bird scales do not significantly overlap, catheterization may be attempted between scutes. The cranial tibial and metatarsal artery offer advantages over other locations recommended for arterial catheterization in that the leg arteries are much easier to secure and maintain once placed. Care must be observed when inserting the catheter into either the cranial tibial artery or the metatarsal artery, it should not be placed too close to the tibiotarsal tarsometatarsal joint, as movement of

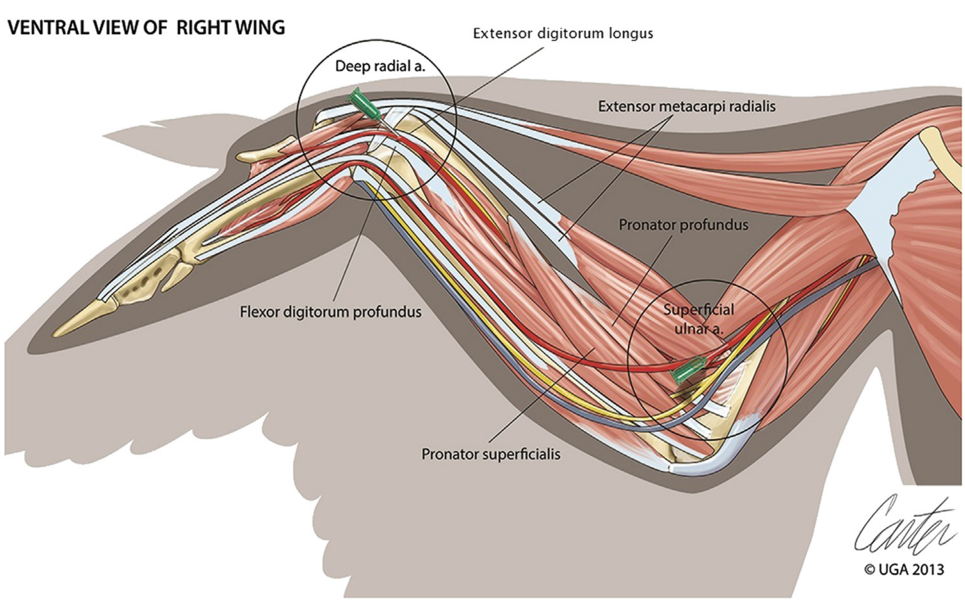

FIGURE 1. The deep radial artery lies between the tendons of the extensor *digitorum longus* and flexor *digitorum profundus* in most birds, whereas the superficial ulnar artery runs medially over the extensor *metacarpi radialis*, pronator *superficialis*, and pronator *profundus* muscles before dipping along the ulna and crossing the carpus to insert into the base of the distal phalanx of the major digit.

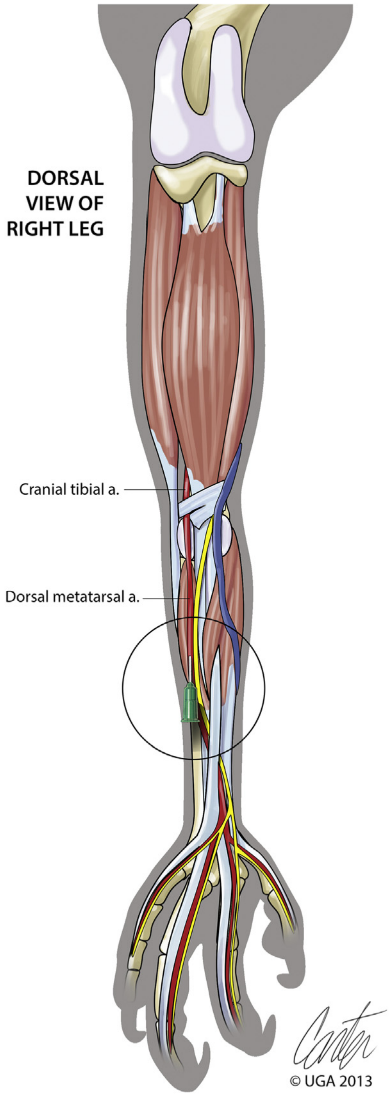

FIGURE 2. Anatomic location of the cranial tibial and metatarsal arteries. The cranial tibial artery crosses the hock dorsolaterally where it becomes the metatarsal artery and travels medially across the tarsometatarsus.

the leg may cause positional occlusion leading to inconsistent readings.

Owing to vessel fragility and the technical difficulty of catheterization, all birds should be anesthetized before catheter placement. For the superficial ulnar and deep radial artery, patients should be positioned in dorsal recumbency with their wings extended.

The insertion site should be aseptically prepared and the catheter should be flushed with heparinized saline (1 U/mL) (Fig. 3). The authors prefer to flush the catheter with heparinized saline before placement, as this facilitates a quicker observation of blood in the needle hub when the catheter enters the artery. Although bird skin is routinely very thin and transparent, it is also very strong and pliable. Without entering the artery, a relief hole or cut down should be made completely through the dermis with a beveled edge of a hypodermic needle just distal to where the arteries are readily palpated (Fig. 4). Arterial location can be confirmed by applying a Doppler transducer over the site and listening for the pulse. Once the artery is palpated and mentally traced, the catheter should be subcutaneously positioned, superficial to the artery, with the bevel up. The catheter needs to be inserted into the artery at a steep angle, and then flattened against the skin surface parallel with the longitudinal axis of the artery, so that the tip of the needle penetrates the most superficial wall of the artery. As both arteries are very superficial, the catheter should be very slowly inserted, watching carefully for blood within the catheter. Once a flash of blood is observed, the catheter should be introduced a few millimeters more, so that the tip of the catheter itself enters the artery. The catheter then can be gently advanced off the stylet to its full length, and the stylet removed (Fig. 5). Advancement should be smooth with little or no resistance. Unless the patient is extremely hypotensive, pulsatile blood flow should be noted from the catheter once the needle stylet has been removed. A T-port or stopcock should be attached to the catheter and the catheter should again be flushed with dilute heparinized saline to ensure its patency (Fig. 6). Owing to the size of these animals, care must be taken to avoid blood loss and a heparin overdose.

The authors have had limited success using adhesive tape in securing both the deep radial or superficial catheters. If a catheter is placed for an anesthetic procedure, tissue glue and/or clear adherent adhesive dressing is sufficient for

FIGURE 3. Materials needed for arterial catheter placement: a 24-gauge catheter, heparinized saline flush, extension set, a piece of tegaderm, and suture material.

FIGURE 4. Catheter placement of deep radial artery. Area is sterilely prepared and the artery is visualized. A cut down or relief hole in the dermis is made using the beveled edge of a needle.

temporary placement. Care must be taken when positioning the animal throughout the procedure. If the catheter is intended to remain while the animal is conscious, the catheter should be sutured in place and incorporated into a figure-of-eight bandage (Fig. 7). Bandaging material should be carefully selected to readily reveal bleeding if the catheter is dislodged. The authors recommend always holding the patient's wing until the arterial catheter is fully secured.

For the cranial tibial or metatarsal artery, catheterization can be performed with the animal in ventral or lateral recumbency while caudally extending its leg. After the insertion site is properly prepared, the thumb and middle finger of one hand locates the artery and the leading edge of same hand can be used to stabilize catheter placement. Once placed however, the catheter is best secured with white adhesive tape.

FIGURE 5. A heparinized saline-prepared intravenous catheter is inserted into the artery, and once a flash is observed it is advanced into the artery.

After the arterial catheter has been successfully placed and secured, it can then be connected to a pressure transducer and a continuous flush mechanism. Ideally, the pressure transducer should be placed as close as possible to the catheter, as this results in the most accurate blood pressure measurements.[32] However, consideration must be made if the catheter is placed in the avian wing. If the transducer is too close to the patient, its weight may act as a fulcrum and dislodge the catheter. Furthermore, the level of the transducers should align with the base or right atrium of the heart. Once the transducer is attached to the monitor, the transducer must be zeroed. Zeroing a transducer is accomplished by opening a 3-way stopcock on the transducer to room air so as to equilibrate the machine with atmospheric pressure and establish a zero reference point. After completion, the monitor then displays the real-time systolic, diastolic, and MAP values for the patient and a series of waveforms. The waveform tracings should be initially assessed for form and consistency.[33] Heart rate values should be compared with values obtained from auscultation to ensure accuracy. In domestic animals, there is a reported 2% to 4% level of inaccuracy associated with direct blood pressure measurements. The blood pressure transducers themselves cause 2% of the inaccuracies, and another 1% to 2% occur because of amplification (e.g., air bubbles in the line). To ensure the most accurate measurements, all air bubbles should be eliminated.[34]

SHORT- AND LONG-TERM MANAGEMENT AND POSSIBLE COMPLICATIONS

Arterial catheterization is generally considered a safe and useful technique that is associated with few serious complications. Although arterial catheterization provides valuable information, managing the catheters may be less than practical in relatively healthy, mobile, or conscious patients. Around-the-clock supervision is imperative for the management of all patients with an arterial line to ensure that an animal does not endanger itself by pulling out its catheter. The authors suggest placement of an Elizabethan collar on the patient and adding tape tabs to the figure-of-eight bandage to serve as a distraction. Circumstances permitting, the authors rarely keep avian arterial lines in place for more than 12 hours. In domestic mammals, it is suggested that the heparinized saline bag should be changed every 24 hours, and the tubing associated with the transducer replaced once every 72 hours.[32] In addition to iatrogenic hemorrhage, there is an increased risk of infection, thromboembolism, and hematoma formation associated with long-term arterial catheter usage. Moreover, frequent administration of heparin can lead to iatrogenic coagulation abnormalities, especially in smaller patients. In human medicine, there is an increased risk of infection and sepsis when an arterial catheter is left in place for greater than 96 hours.[35] Sepsis is also more prevalent when local inflammation is present. Although rare, other complications such as cellulitis, abscess formation, temporary occlusion of the artery, nerve paralysis, suppurative thromboarteritis, arteriovenous fistulas, and pseudoaneurysm have been reported in domestic animals. Fluids and medications should never be administered via the arterial catheter because of the potential risk of vascular damage and subsequent tissue necrosis.[7] After removing the catheter, pressure should be applied over the insertion site with dry cotton swabs or gauze. It usually takes 3 to 5 minutes for complete hemostasis after catheter removal. Patients should be monitored until no bleeding is observed.

WAVEFORM INTERPRETATIONS

Arterial blood pressure is determined by cardiac output and systemic vascular resistance and is a primary determinant of cerebral and coronary perfusion. Arterial blood pressure can be further delineated into 3 components: systolic, diastolic, and mean arterial blood pressures. Systolic blood pressure is primarily measured by stroke volume and arterial compliance. Diastolic blood pressure is mainly determined by systemic vascular resistance and heart rate. Mean blood pressure is the average pressure consisting of half of the area of the pulse-pressure waveform. Physiologically, mean blood pressure is the most significant value because it denotes the average driving pressure for organ perfusion.[5]

The relationship between systolic blood pressure and mean arterial blood pressure is variable and is reliant on the shape and amplitude of the pulse-pressure waveform. The pulse-pressure waveform is largely a reflection of stroke volume and vessel size. Assessment of the waveform is critical for evaluating cardiac function. Peak left ventricular ejection occurs during the highest point on the waveform and is associated with the systolic fraction. The descending portion of the waveform is associated with a drop in pressure. Within the first half of the waveform descent, a dicrotic notch may be visible. The dicrotic notch indicates the closure of the aortic valve and represents the beginning of diastole. The lowest point of the waveform represents the end of diastole before the next heartbeat is exhausted (Fig. 8).[5]

FIGURE 6. A T-port and stopcock is attached to the catheter, and it flushed with heparinized saline.

Analysis of the arterial waveforms can provide insight into the nature of the blood pressure abnormalities. A rapid decline in the downward stroke of the waveform frequently indicates rapid runoff of blood, and hence decreased systemic vascular resistance, whereas a slow decline in the downward stroke of the arterial waveform may indicate increased systemic vascular resistance. Small, narrow pulse-pressure waveforms are frequently observed with small stroke volumes or vasoconstriction. Small stroke volume has also been noted with hypovolemia, poor heart function, tachycardia, or ventricular arrhythmias. Tall, wide pulse-pressure waveforms in domestic animals are commonly associated with sepsis. Tall, narrow waveforms are observed in animals with

FIGURE 7. Suture and a piece of tegaderm are used to secure the catheter.

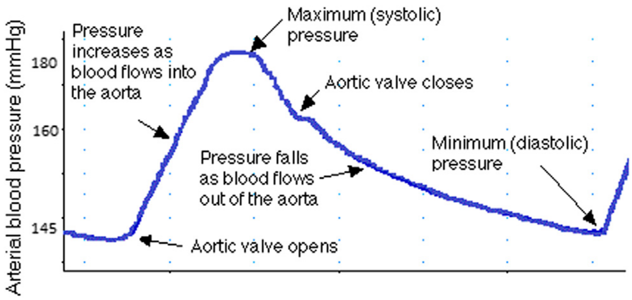

FIGURE 8. A normal avian blood pressure waveform.

patent ductus arteriosus or arteriovenous fistula, or during cardiopulmonary resuscitation.[5]

Falsely decreased systolic and elevated diastolic values may be present with a dampened waveform. Dampening can occur because of a poor state of perfusion. Waveform dampening can be evident in patients with severe peripheral vasoconstriction either because of severe hypovolemia or through the use of high doses of vasopressor agents. Waveform dampening is commonly associated with cardiac arrhythmias, such as intermittent premature ventricular contractions, and occurs in conjunction with abnormal MAPs. Dampening waveforms have also been linked with air bubbles, blood clots, excessive blood, or catheter or tube line occlusion. Furthermore, arterial spasms can lead to noticeable dampening. Although the dampening produces an abnormal waveform, the mean arterial blood pressure is generally correct in these situations. In all cases of dampening, the line and catheter should be checked to assess for mechanical anomalies.[36]

Amplification can also occur and usually represents the reflections of the waveform from a peripheral catheter. These reflections amplify systolic pressures, resulting in falsely elevated values. Amplification is also observed in arteries that are noncompliant, such as those recorded in geriatric patients or patients with atherosclerotic and arteriosclerotic lesions.[36]

Care must be taken when sudden dramatic changes in blood pressure are noted. If the problem does not seem to be artificial, a sudden change in pressure can indicate that cardiac arrest has occurred, or that it is imminent and immediate action is required.[35]

BLOOD GASES

Arterial blood gas is the gold standard for assessing an animal's acid-base status, ventilation, and tissue perfusion. It provides essential physiologic information for patients with critical illness or respiratory disease and is vital in the correction of any metabolic or respiratory disorders. However, there are limited amount of data available for avian blood gas values. Few arterial blood gas studies have been performed on a variety of avian species (Tables 2 and 3).[37,38] Most of the avian blood gas studies were used to compare the effects of anesthetic conditions, exercise, or changes in altitude in the bird subjects compared with their baseline values. Point-of-care blood gas analyzers directly measure the pH, partial pressure of oxygen (PO_2), and partial pressure of carbon dioxide (PCO_2) and can then be used to calculate hemoglobin saturation with oxygen, bicarbonate concentration (HCO_3), total CO_2 concentration, and base excess of the extracellular fluid.[37]

Blood pH represents the overall balance of all the acids and bases within the body. It is influenced by respiratory (PCO_2) and metabolic (HCO_{3-}) components. PCO_2 provides essential information regarding ventilation. Partial pressure of carbon dioxide >45 mm Hg in most domestic species is associated with hypoventilation. Common causes of hypoventilation include decreased respiratory brain activity (e.g., anesthesia, sedation, trauma, and edema) or improper flow of air through the airways (e.g., upper airway obstruction, lower airway obstruction, and a mass in the coelomic cavity compressing the air sacs). Hyperventilation is characterized by decreases in PCO_2, which can lead to respiratory alkalosis defined as $PCO_2 <$ 35 mm Hg in most domestic species. Common causes of hyperventilation include hypoxemia, pulmonary disease, pain, anxiety, excessive manual or mechanical ventilation, or compensatory mechanisms for metabolic acidosis.[37]

TABLE 2. Acid-base status in the avian patient using a portable point-of-care analyzer.[37] Arterial blood gases and pH are from nonanesthetized birds breathing room air

Bird Species	PO_2	PCO_2	pH
White leghorn chicken	82	33.0	7.52
Muscovy duck	96.1	36.9	7.46
Pekin duck	100	33.8	7.48
Emu	99.7	33.8	7.45
Bar-headed goose	92.5	31.6	7.47
Herring gull	–	27.2	7.56
Red-tailed hawk	108	27.0	7.49
Pigeon	77.1	40.9	7.50
Burrowing owl	97.6	32.6	7.46

Parameter	Units	Mean	SD	Minimum	Maximum
iCa	mmol/L	0.8	0.28	0.34	1.4
K	mEq/L	3.5	0.53	2.8	4.9
Na	mEq/L	147.4	2.2	141	150
pH	$\log_{10}$ (1/H)	7.452	0.048	7.343	7.552
PCO_2	mm Hg	22.1	4	14.6	29.8
PO_2	mm Hg	98.1	7.6	85	113
Ht	%	38.7	6.2	24	50
Hb	g/dL	13.2	2.1	8.2	17
HCO_3	mmol/L	14.8	2.8	9.5	21
SO_2	%	96.2	1.1	94	98
BE	mmol/L	−7.9	3.1	−15.1	−1
Respiration rate	breaths/min	82	33	32	150
Temperature	°C	41.8	0.6	40.2	43

TABLE 3. Blood and electrolyte values for nonanesthetized Amazon parrots (*Amazona aestiva*).[38]

SD, standard deviation; BE, base excess of the extracellular fluid.

The metabolic contribution to the acid-base balance can be assessed with the HCO_3 and the base excess of the extracellular fluid. A study in Amazon parrots reported a baseline mean HCO_3 reference range of 14.8 ± 2.8 mmol/L. Values that are below this range may indicate metabolic acidosis, whereas values greater than this range indicate metabolic alkalosis. Metabolic acidosis can be caused by increased generation of acidic metabolic products such as lactate or ketones or the inability of the kidneys to eliminate those products. As the acid increases, it becomes buffered, resulting in a subsequent decrease in the HCO_3 levels. Metabolic acidosis can also occur through a direct loss of bicarbonate from the gastrointestinal tract or kidneys.[39] Partial pressure of oxygen gives a good indication of tissue perfusion. Tissue hypoxemia occurs when PO_2 values are below 80 mm Hg. Presence of persistently hypoxemic conditions can be life threatening, and a PO_2 value below 60 mm Hg warrants immediate therapeutic intervention.[37]

Arterial catheterization is often needed to obtain serial blood gas samples. A small amount of blood should first be drawn from the catheter as a "waste sample." This ensures that any heparinized saline in the catheter is removed and does not dilute or contaminate the sample. With small patients, care must be taken to not collect an excessive amount of blood. The authors usually disconnect the catheter from the line and blood pressure transducer and collect only 0.2 mL as a waste sample. Fortunately, most blood gas analyzers (e.g., i-STAT, Abaxis, Union City, CA USA) require 0.06 to 0.2 mL of blood to obtain results. All air bubbles should be removed from the blood in the syringe after which the sample is capped and immediately processed. The arterial catheter and T-connector are then flushed with a small amount of heparinized saline to prevent clot formation. Results from blood gas analysis can be altered by several factors, including the higher core body temperature found in birds. Blood gas analyzers have temperature conversion formulas within their software to correct for core body temperature differences; however, to the authors' knowledge, there have been no published studies determining the accuracy of this equation in avian species.[37]

Blood gas analyzers have become a useful tool in the diagnosis and treatment of avian acid-base conditions. Respiratory acid-base abnormalities should be treated by eliminating the underlying disease and providing appropriate ventilation therapy, if required. Most metabolic disorders can be corrected with appropriate fluid therapy. However, sodium bicarbonate may be indicated in patients with metabolic acidosis associated with renal insufficiency or severe metabolic acidosis despite recommended fluid therapy administration.[37]

HYPOTENSION

As previously stated, the definition of true hypotension in birds is somewhat vague, and further studies are needed to establish a better understanding between blood pressure and tissue perfusion. However, decreases in blood pressure because of factors such as hypovolemia, hemorrhage, concurrent disease, inflammation, sepsis, and adverse effects of anesthesia should be treated in all patients. Hypotension treatment should be based on underlying etiology, but it usually consists of decreasing anesthetic inhalant, volume resuscitation (crystalloid or colloids), or vasopressors.[20] The daily maintenance fluid rate for birds is approximately 60 to 100 mL/kg/d. Crystalloid fluids are frequently administered intravenously for volume support, maintenance, and rehydration. In healthy anesthetized patients, replacement fluid rates of 10 mL/kg/h of crystalloids are often recommended. For hypotensive birds, fluid boluses of 10 to 20 mL/kg over a few minutes are generally well tolerated. The authors have used shock dosages of 90 mL/kg of crystalloid fluid therapy over 5 to 7 minutes for severely compromised birds, but this does not suggest the use of such high dosages unless the animal is severely compromised and the underlying disease condition is identified.[23] Colloidal fluids (e.g., hetastarch, hypertonic saline, and whole blood) have also been used to treat hypotension due to significant hypovolemia. A study in critically ill raptors reported that boluses of hetastarch administered at doses of 10 to 15 mL/kg every 8 hours for less than or equal to 4 treatments appeared to be well tolerated.[40] Alternatively, dosages less than or equal to 20 mL/kg/d are suggested. For severe hemorrhage, whole-blood transfusions should be provided. If whole blood is not available, it has been recommended to use crystalloid fluids at 3 times the volume of blood loss.[30]

There have only been a few studies on the effects of vasopressor agents in birds. A study performed on anesthetized red-tailed hawks (*Buteo jamaicensis*) showed that the administration of norepinephrine at a rate of 0.4 to 5 μg/kg/min caused an increase in blood pressure and hypertension.[10] In another study, performed on anesthetized hypotensive Hispaniolan Amazon parrots, a constant rate infusion of dobutamine at 5, 10, and 15 μg/kg/min and dopamine at 5, 7, and 10 μg/kg/min increased systemic blood pressures to normotensive levels.[25]

HYPERTENSION

Normotension is significantly higher in most avian species than in mammals. In the authors' experience, systolic direct blood pressure values >240 mm Hg in conscious and anesthetized birds are considered to indicate hypertension. An increase in systemic vascular resistance is a common denominator in most hypertension cases. Organ blood flow can be maintained at a relatively constant level even though there can be significant variations in perfusion; however, if systemic pressures exceed autoregulatory ranges, targeted organ damage can occur. Hypertension can result in endothelial damage, platelet and fibrin deposition, ischemia, fibrinoid necrosis, and/or hemorrhage. Hypertensive animals can also display neurological, cardiovascular, or renal abnormalities, with the eyes, brain, heart, and kidneys being the most commonly affected organs.[41]

Hypertension is classified as either primary or secondary hypertension. Although quite common in humans, primary hypertension has been rarely documented in veterinary medicine, and its prevalence is unknown in birds. Secondary hypertension occurs as a result of other conditions that affect the kidneys, arteries, heart, or endocrine systems. Uric acid monitoring with or without endoscopic renal biopsy may help rule out renal causes of hypertension. Atherosclerosis is thought to be the most common vascular disease in birds.[2,3] Atherosclerosis causes thickening of the arterial wall associated with lipid deposition, which leads to increases in vascular resistance, a decrease in the vascular lumen, and ultimately systemic hypertension. Furthermore, in human studies, systemic hypertension has been reported as an underlying cause of endothelial damage, inflammation, and the development of atherosclerosis.[42] Hypertension has also been linked to an increased risk of ischemic heart disease, stroke, peripheral vascular disease, and other cardiovascular disorders, including heart failure, aortic aneurysm, and pulmonary embolism.[43] Evidence in human medicine suggests that reduction of the blood pressure by 5 mm Hg can decrease the risk of stroke by 34% and of ischemic heart disease by 21% and reduce the likelihood of dementia, heart failure, and mortality from cardiovascular disease.[42]

Treatment for high blood pressure consists of dietary changes, exercise, and administration of antihypertensive agents. Long-term salt and caloric restrictions are beneficial in lowering blood pressures in human patients. There are many antihypertensive agents, including diuretics, angiotensin-converting enzyme (ACE) inhibitors, calcium channel blockers, β-blockers, and other vasodilators.[44]

Diuretics reduce blood pressure by the decreasing extracellular fluids. Diuretics are commonly used in human patients with hypertension, but they have not been particularly beneficial in veterinary medicine for decreasing blood pressure in animals (owing to primary hypertension being diagnosed more often in humans vs animals). Diuretics are contraindicated in hypertensive patients who are dehydrated or have metabolic imbalances.[43] β-Adrenergic receptor blockers are thought to decrease blood pressure by reducing heart rate and cardiac contractility, thereby decreasing cardiac output.[44,45] β-Blockers are considered the treatment of choice in hypertensive cats that are diagnosed with hyperthyroidism.[7] ACE inhibitors control blood pressure by blocking the conversion of angiotensin I to angiotensin II, which reduces the production of potent vasoconstrictors and decreases the secretion of antidiuretic hormone and aldosterone. This ultimately promotes sodium and water excretion and leads to a decrease in extracellular fluids and lower blood pressures. A study performed on hypertensive cats with chronic renal failure showed that benazepril administration resulted in a decrease of both glomerular capillary and systemic arterial blood pressure.[41] Calcium channel blockers decrease blood pressure by promoting vasodilation and by preventing an increase in cytosolic calcium within the vascular endothelial cells, inhibiting vasoconstriction. Although these agents do cause preglomerular vasoconstriction, which can decrease the glomerular filtration rate, they also appear to have renoprotective properties. Calcium channel blockers are thought to prevent renal injury by limiting renal growth, by reducing mesangial entrapment of macromolecules, and by attenuating the mitogenic effects of diverse cytokines and growth factors.[43] Studies in cats and dogs have shown that amlodipine, a long-acting dihydropyridine calcium antagonist, is effective in controlling system hypertension. Owing to its efficacy and low rate of adverse effects, it is considered the antihypertensive drug of choice in most domestic mammals.[6,46] Other vasodilators such as phenothiazine and other derivatives have been used to control hypertension. In hypertensive cats, the administration of hydralazine resulted in a reduction in systolic blood pressure to normal values within 15 minutes.[43] In humans with occlusive artery disease, administration of peripheral vasodilators (e.g., isoxsuprine) are beneficial to patients with intermittent limb pain, weakness, and lameness, particularly those diagnosed with femoral or lower leg vascular obstructions. Isoxsuprine treatment of clinical signs associated with presumptive atherosclerosis in a yellow-naped Amazon parrot (*Amazona auropalliata*) resulted in vascular smooth muscle relaxation, possibly through α- and β-adrenergic receptor stimulation, although the exact mechanism of action is unknown.[47]

One must remember that there is little information regarding the diagnosis and treatment of cardiovascular disease in birds. Most of the current published veterinary medical information consists of anecdotal case reports. Only a limited number of scientific studies have addressed the use of cardiovascular drugs in avian species. Of the few studies performed, most have used galliformes. One study found that in turkeys, β-blockers, such as oxprenolol administered at doses of 2 mg/kg, orally, once daily, had a protective effect against the development of atherosclerotic plaques and arterial wall hypertrophy.[48] A study in broiler chickens reported that repeated administrations of propranolol intramuscularly at 4 to 8 mg/kg/d caused a reduction of blood pressure by 19.1 ± 3.0 mm Hg and heart rate 76 ± 6 beats per minute.[49] Furthermore, a study using pranidipine, a new dihydropyridine calcium antagonist, in turkeys with experimentally induced dilated cardiomyopathy resulted in significantly smaller left ventricular dimensions compared with controls and did not cause any adverse effects to any of the birds used in the investigation.[50] Another research project using broiler chickens showed that the administration of oral diltiazem at a dose of 15 mg/kg every 12 hours greatly reduced low temperature–induced pulmonary hypertension and vascular remodeling.[21] Another study in turkeys reported that the ACE inhibitor captopril prevented elevations of systemic blood pressures when animals were administered angiotensin I.[51] Enalapril administered at a dose of 5 mg/kg once a day was also used to treat a lovebird with pericardial effusion for 11 months without any reported adverse side effects.[52] Anecdotal reports in parakeets and an Amazon parrot administered benazepril at a dose of 0.5 mg/kg, orally twice daily, controlled hypertension in these animals for several months without adverse side effects.[19] Other antihypertensive agents such as β-adrenergic agonists have been anecdotally administered to birds.[53] Isoxsuprine administered orally at a dose of 10 mg/kg once a day has been used to treat the clinical signs of ataxia and lethargy associated with presumptive atherosclerosis in a yellow-naped Amazon parrot with no adverse side effects. After 1 year of isoxsuprine therapy, treatment was discontinued for approximately 10 months. Clinical signs recurred and again resolved when treatment was resumed. Tibiotarsal blood pressure measured in the follow-up period, while being treated with isoxsuprine, remained in the range of 160 to 190 mm Hg.[47]

Most human patients require more than one therapeutic agent to control systemic hypertension. The Joint National Committee on High Blood Pressure suggests treating people with 2 or more drugs when blood pressure exceeds 20 mm Hg more than normal systolic or 10 mm Hg more than normal diastolic pressures.[44] For the treatment of hypertension in birds, the authors suggest a combination of renin-angiotensin system inhibitors (such as an ACE inhibitor) and calcium channel blockers or a diuretic.

CONCLUSION

Although arterial catheterization of birds is initially a difficult technical procedure, direct blood pressure measurement is the gold standard for diagnosing hypotension or hypertension. Direct blood pressure measurement should ideally be used in all critically ill or anesthetized avian patients as well as with patients when hypertension or atherosclerotic disease is suspected. Direct blood pressure measurement provides veterinarians dynamic information, enabling accurate monitoring and treatment recommendations for numerous cardiogenic and cardiovascular disease and conditions.

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