CHAPTER

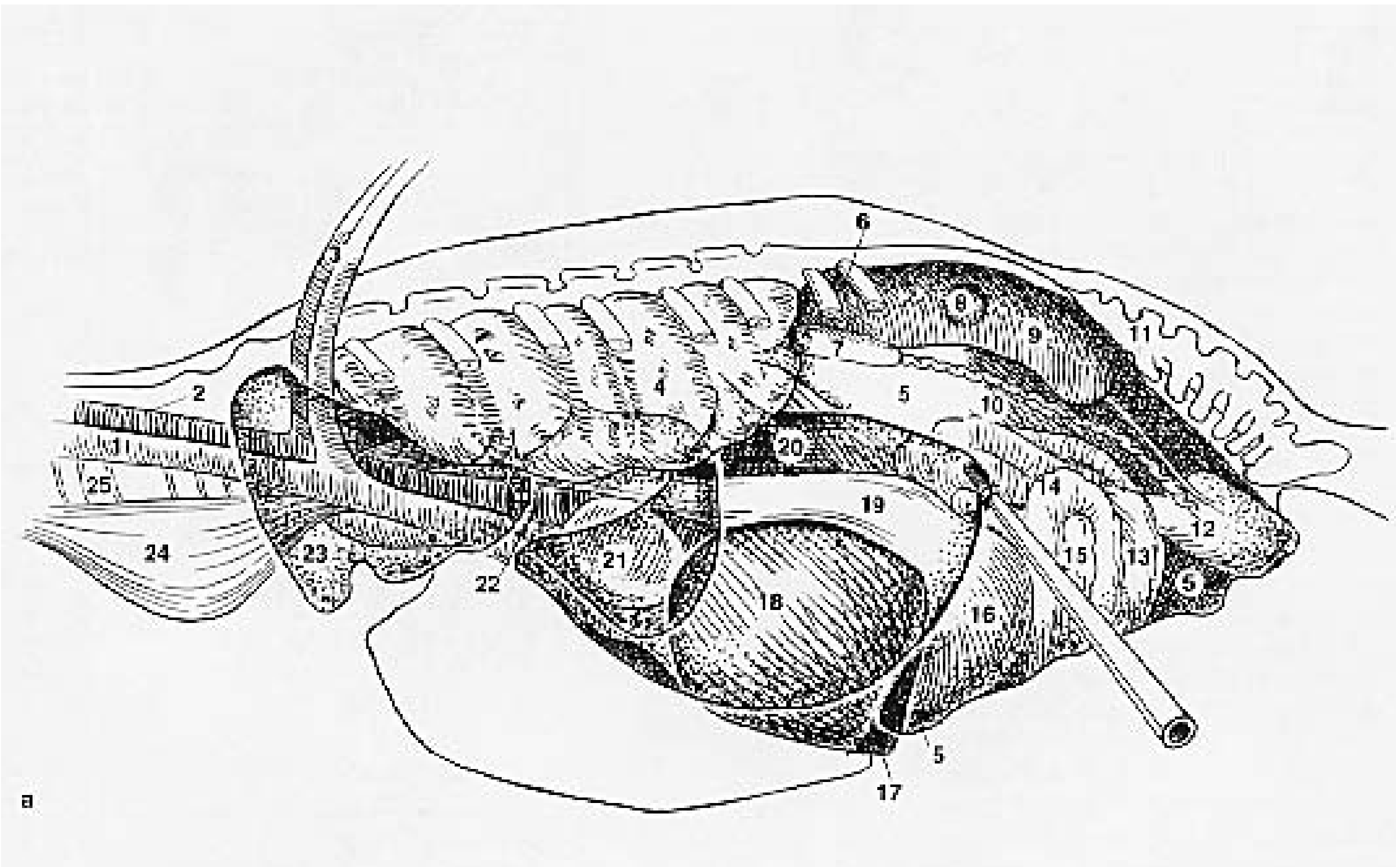
13

ENDOSCOPIC EXAMINATION AND BIOPSY TECHNIQUES

Michael Taylor

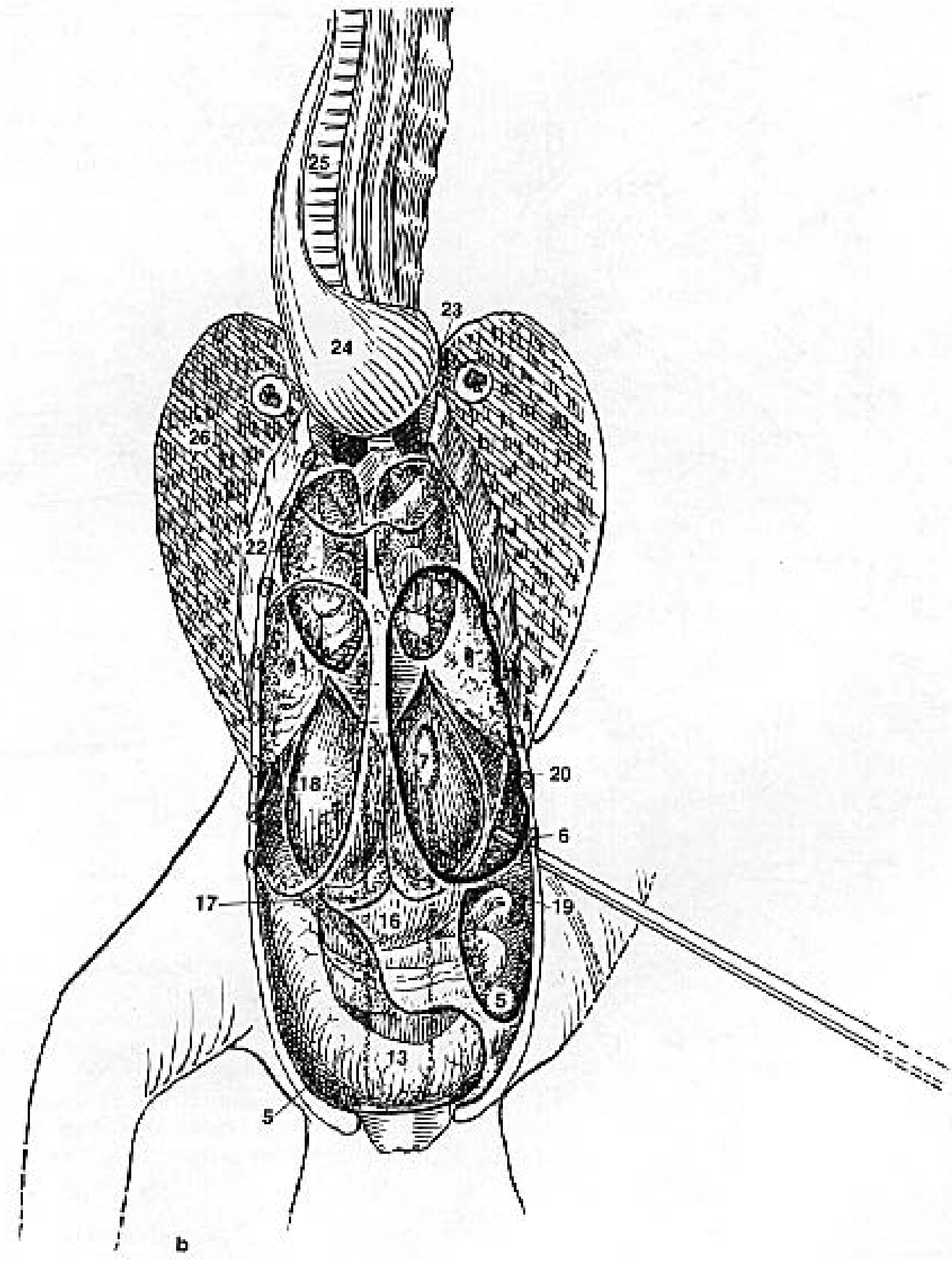
The development of a rigid rod-lens system and the perfection of fiberoptic cables in the late 1950's and early 1960's heralded the modern age of endoscopy in human medicine. A unique lens design[9] allowed for improved light transmission in small diameter telescopes. Over the next decade, various rigid endoscopes were introduced into human gynecology, orthopedics and otolaryngology. By the middle 1970's veterinarians were employing these endoscopes in animal species, and the concept of rigid endoscopy was introduced to avian practitioners.[2,27]

A growing interest in aviculture, particularly of psittacine birds, must also be credited with stimulating the field of avian endoscopy. Endoscopic determination of gender (surgical sexing)[22] has become an integral part of the captive management of many avian species. Birds are ideal subjects for endoscopic examination due to the unique design of their respiratory system, which provides extensive pneumatization of the coelom. A variety of diagnostic uses for endoscopy in birds has previously been described;[2,8,19,21] however, the greater benefits of this technology have hardly been explored. New developments in equipment and techniques are certain to increase the value of endoscopy to avian veterinarians.



1) internal carotid artery
2) jugular vein
3) subclavian vessels
4) lung
5) abdominal air sac
6) eighth rib
7) gonad (in this case a testicle)
8) oscoxae
9) kidney
10) vas deferens
11) ureter
12) cloaca
13) colon
14) duodenum
15) pancreas
16) ventriculus
17) ventral hepatic peritoneal cavity
18) liver
19) proventriculus
20) caudal thoracic air sac
21) heart
22) cranial thoracic air sac
23) clavicular air sac
24) crop
25) trachea
26) pectoral muscle

FIG 13.1 a) An artist's impression of the lateral view of a bird showing anatomic structures of importance when endoscope is in position 6 (see Figure 13.2).



1) internal carotid artery	8) oscoxae	15) pancreas	22) cranial thoracic air sac
2) jugular vein	9) kidney	16) ventriculus	23) clavicular air sac
3) subclavian vessels	10) vas deferens	17) ventral hepatic peritoneal cavity	24) crop
4) lung	11) ureter	18) liver	25) trachea
5) abdominal air sac	12) cloaca	19) proventriculus	26) pectoral muscle
6) eighth rib	13) colon	20) caudal thoracic air sac	
7) gonad (in this case a testicle)	14) duodenum	21) heart	

b) An artist's impression of the VD view of a bird showing anatomic structures of importance when performing endoscopy from various entry sites: When the scope is introduced through entry site 6 (see Figure 13.2), it enters the caudal thoracic air sac. On the VD view it may appear as though the scope goes through the abdominal air sac as well. The abdominal air sac actually forms a backwards C positioned dorsal and ventral to the caudal thoracic air sac (see Anatomy Overlay). In some species, the right and left abdominal air sacs may be more symmetrical than shown.

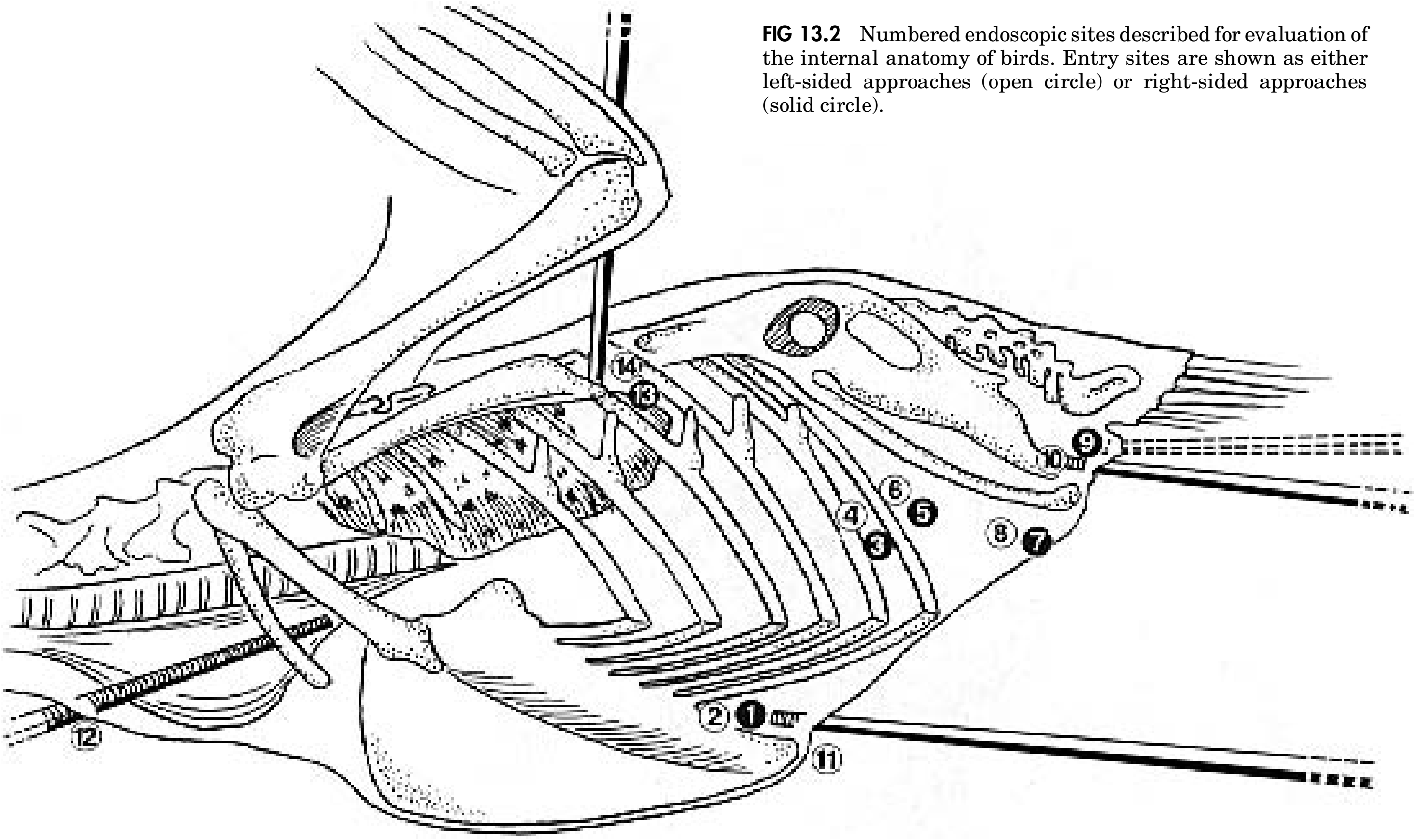


FIG 13.2 Numbered endoscopic sites described for evaluation of the internal anatomy of birds. Entry sites are shown as either left-sided approaches (open circle) or right-sided approaches (solid circle).

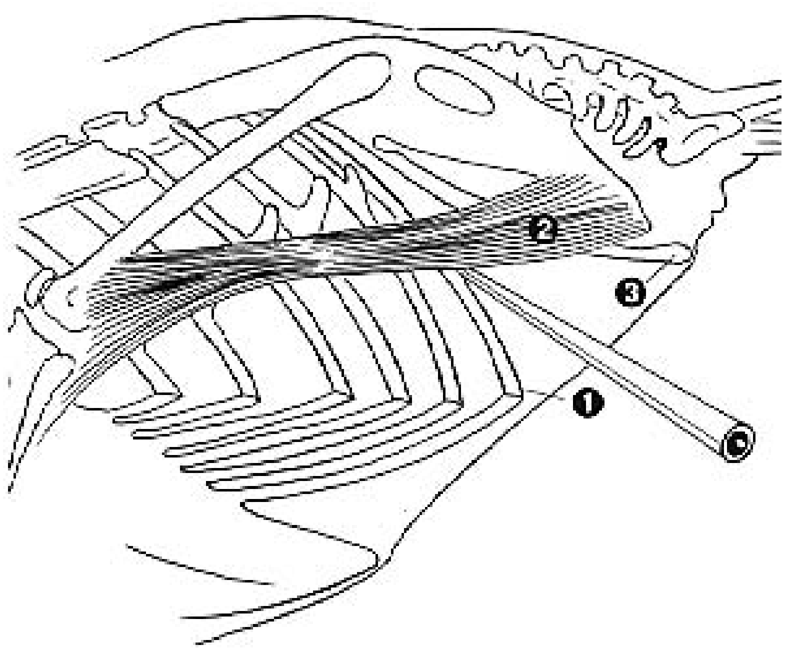


FIG 13.3 Left flank approach for endoscopic evaluation of the bird. The leg is pulled cranially and the entry site is at the junction of the caudal edge of the 1) eighth rib and the 2) flexor cruris medialis muscle. 3) The pubic bone serves as an additional landmark. This approach is listed as entry site 6 in Figure 13.2.

Endoscopic laparotomy can be performed from either the right or left side of a bird, and 14 different approaches have been described. These approaches are depicted in Figure 13.2. Site 4, located between the seventh and eighth ribs, is frequently used for endoscopic evaluation of the gonads; however, an entrance point through the left flank (site 6, Figures 13.2, 13.3) just ventral to the flexor cruris medialis muscle and caudal ventral to its intersection with the vertebral portion of the eighth rib and the pubic bone is a site that provides better visualization of many abdominal structures.

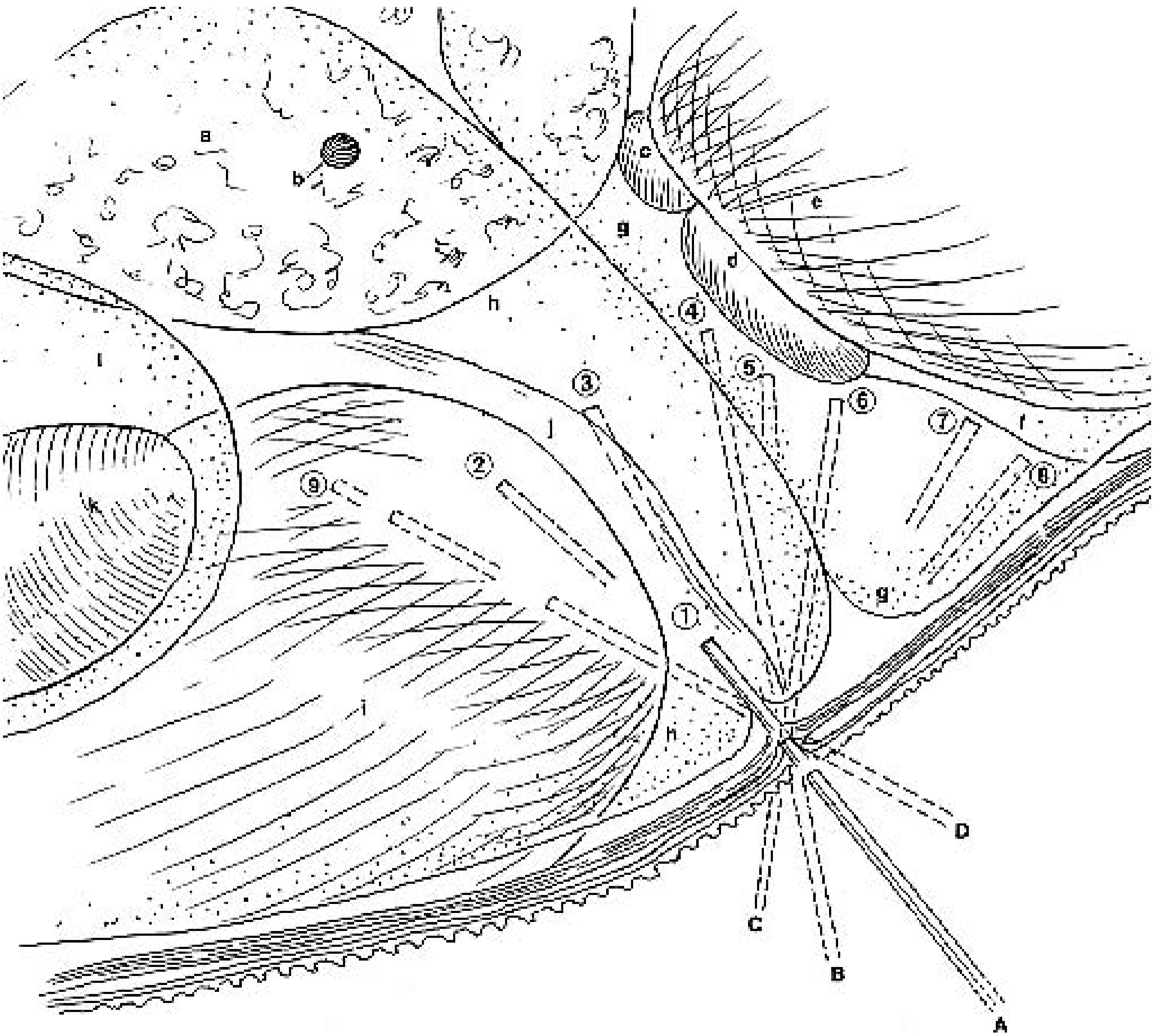


FIG 13.4 An artist's rendition of the anatomic features that are visible when the endoscope is placed in different directions and at different depths from entry site 6 (Figure 13.2). By matching the angle and depth of the endoscope, the endoscopist can develop an insight into the relative position of organs as viewed from entry site 6. The views are divided into four angles (A,B,C,D) and depths (1 through 9). Each color endoscopic picture has a corresponding angle and position marker to help the endoscopist envision the anatomic relationship of the endoscopic view. Thus, if the scope is oriented to B-4, the gonad, adrenal gland and kidney would be in view. Structures that will be used for orientation in the various endoscopic pictures include: a) lung b) ostium of the cranial thoracic air sac c) adrenal gland d) gonad e) kidney f) ureter, oviduct, vas deferens area g) abdominal air sac h) caudal thoracic air sac i) liver j) proventriculus k) heart and l) cranial thoracic air sac.

Endoscopic Examination and Biopsy Techniques

Understanding the relationship of structures is critical to effective endoscopy. This approach to endoscopic anatomy will guide the clinician through the evaluation of thoracoabdominal structures that can be viewed from various entrance points to the abdominal cavity. (All color photographs in this section © 1994 by Michael Taylor.)

Color 13.1
The left abdominal wall has been removed from an Amazon parrot. Note the tiered effect of the cranial thoracic (open arrows), caudal thoracic (arrows) and abdominal air sacs (a). The intestinal peritoneal cavity (IPC) has been infused with red dye. Other prominent organs include the lung (lu), heart (h), liver (li) and proventriculus (p).

Color 13.2
A bird is placed in right lateral recumbency with the leg extended cranially to show the insertion point to entry site 6 (see Figure 13.2). Dotted lines mark the caudal edge of the eighth rib (r), the flexor cruris medialis muscle (m) and the pubic bone (p). The entrance site is at the junction of the eighth rib and the flexor cruris medialis muscle.

Color 13.3
Endophotograph of entry site 6 to show the eighth rib (r), ventral border of the flexor cruris medialis muscle (m) and the penetration point in the lateral abdominal wall.

Color 13.4
(Position A-1 see Figure 13.4) Immediately after entering the abdominal cavity of an Amazon parrot, the caudal thoracic air sac can be visualized. The air sac should be transparent with minimal vascularity. The proventriculus (p) is ventral to the endoscope. The medial wall of the caudal thoracic air sac (a) becomes contiguous with the lateral wall of the abdominal air sac.

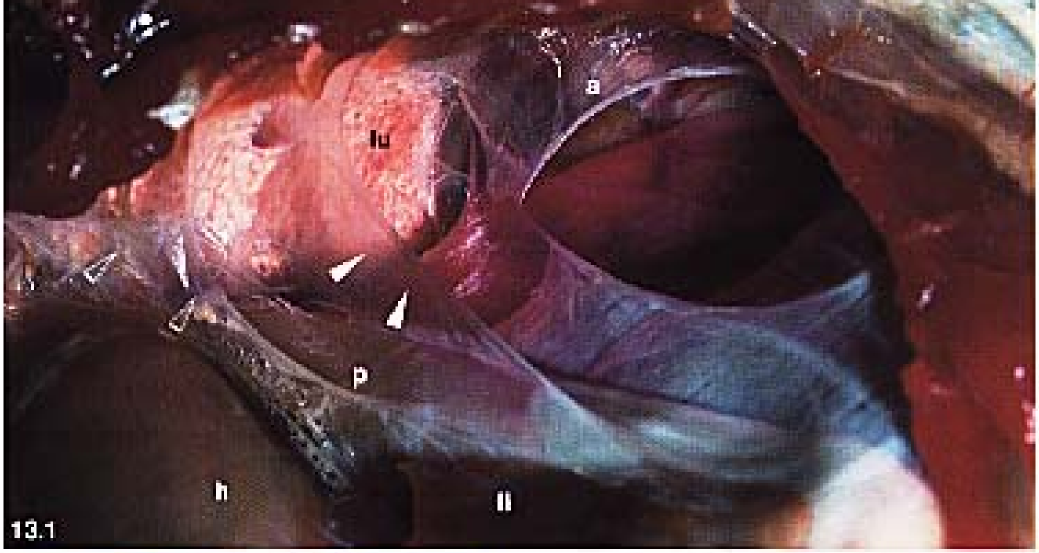
Color 13.5
(Position A-2 see Figure 13.4) Normal caudal thoracic air sac of an Amazon parrot. In this view, a clear, unobstructed view of the ostium (o) of the caudal thoracic air sac indicates that the tip of the endoscope is within this air space. Also visible are the dorsal edge of the left liver (li), lung (lu), proventriculus (p) and the confluent wall of cranial thoracic and caudal thoracic air sac (open arrow). The proventricular arteries are clearly visible (arrow).

Color 13.6
(Position A-3 see Figure 13.4) Normal organs and air sacs in an Amazon parrot. Ostium (o), lung (lu), proventriculus (p), liver (li), confluent wall of cranial and caudal thoracic air sacs (open arrow) and confluent wall of caudal thoracic and abdominal air sacs (arrow).

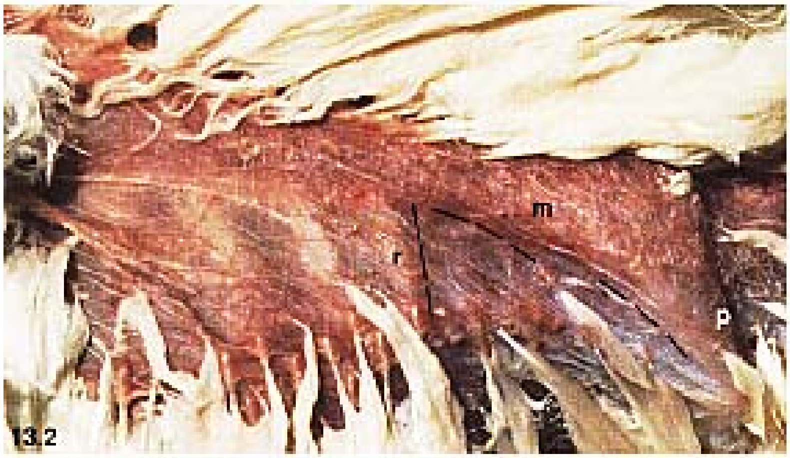
Color 13.7
(Position B-4 see Figure 13.4) Mature melanistic left testicle (t) of a Goffin's Cockatoo. Also visible are the left adrenal gland (a), ilium (i), cranial pole of the left kidney (k), left common iliac vein (arrow) and aorta (open arrow).

Color 13.8
(Position B-4 see Figure 13.4) Normal immature testicle (t) of a Quaker Parakeet. Also visible are the left adrenal gland (a), right and left common iliac veins (arrows) and the caudal vena cava (open arrow).

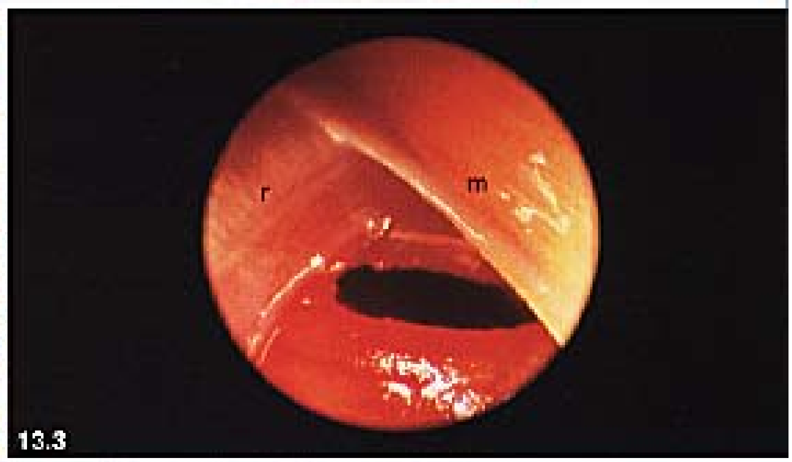
Color 13.9
(Position B-4 see Figure 13.4) Endoscopic view of the kidney (k) and immature melanistic ovary (o) of a six-month-old Blue and Gold Macaw. Note the sulci and gyri. Vessels are seen through the abdominal air sac in the peritoneal membrane overlying the gonads (arrows).



13.1

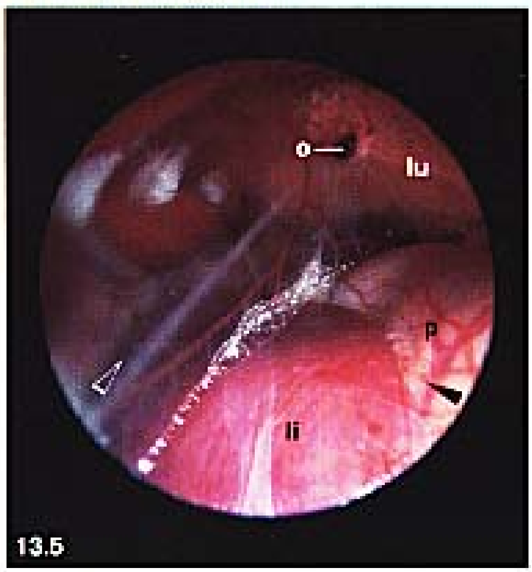


13.2

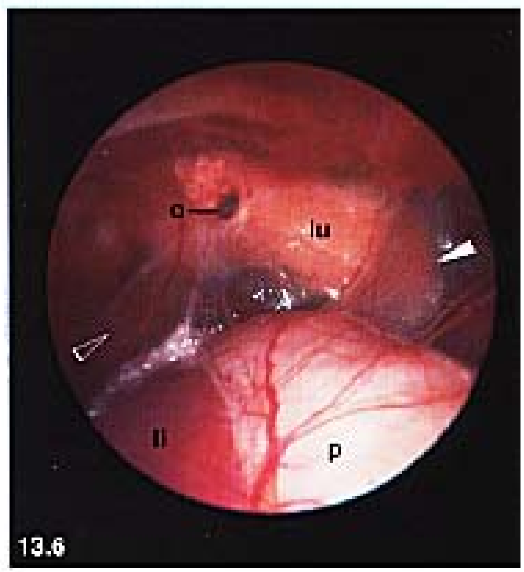


13.3

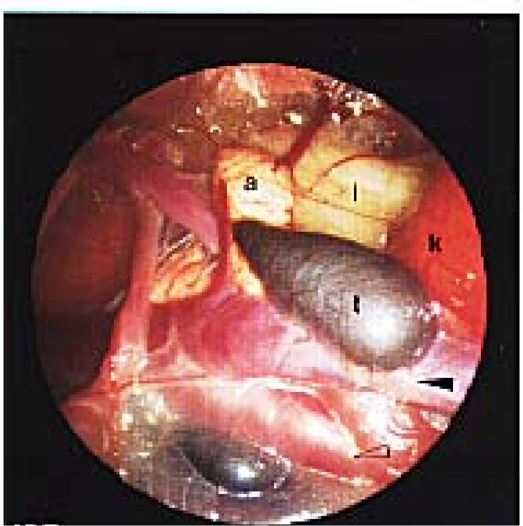
13.4

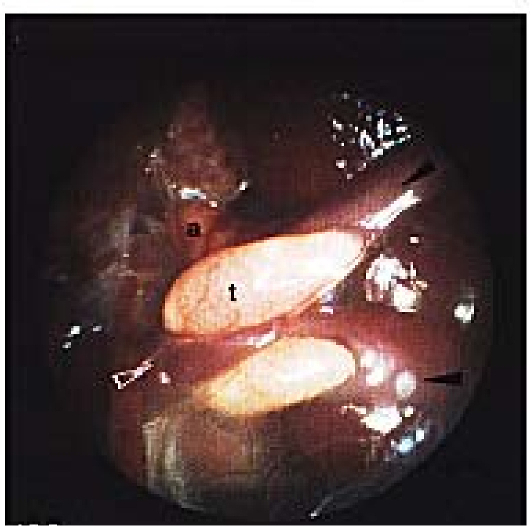


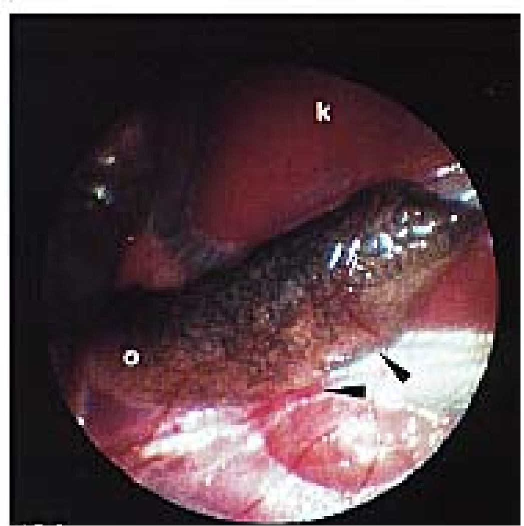
13.5

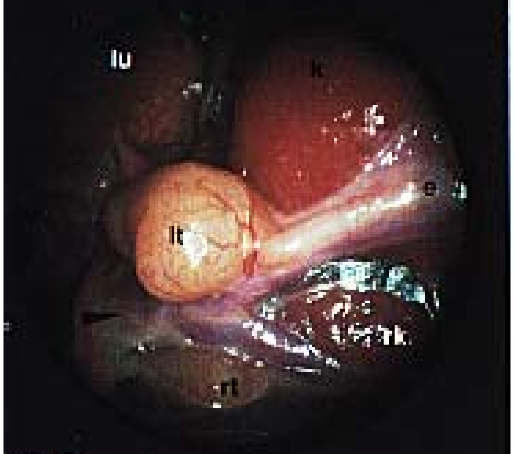


13.6

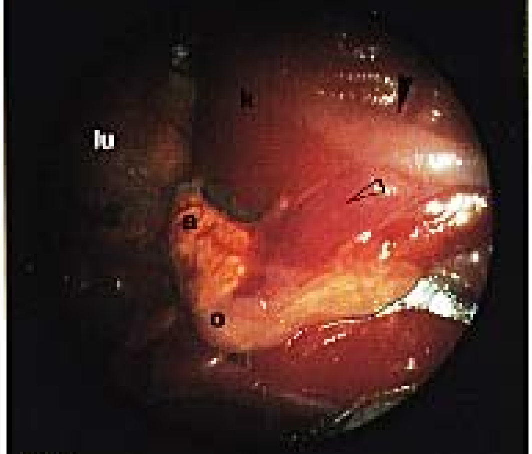




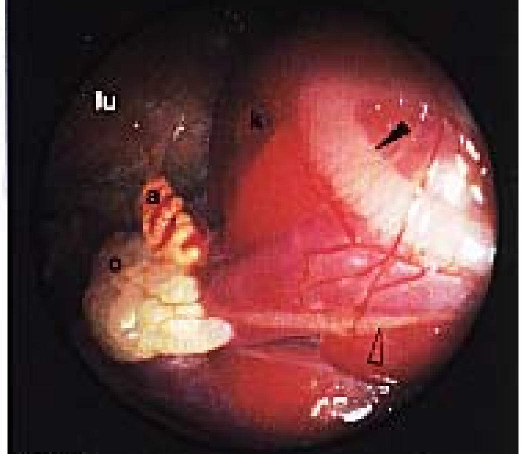




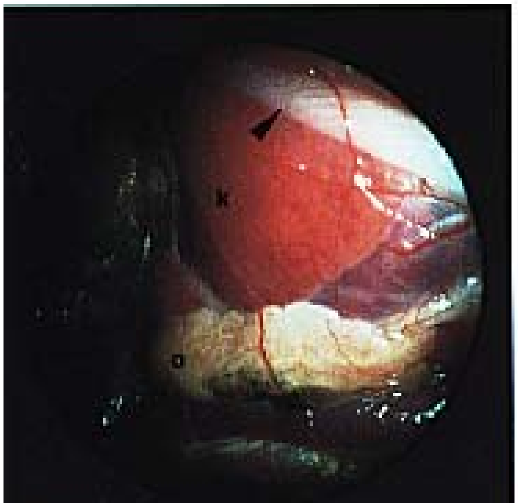
13.10



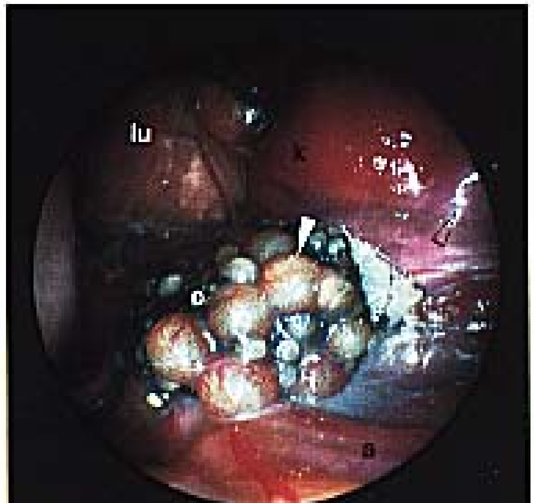
13.11



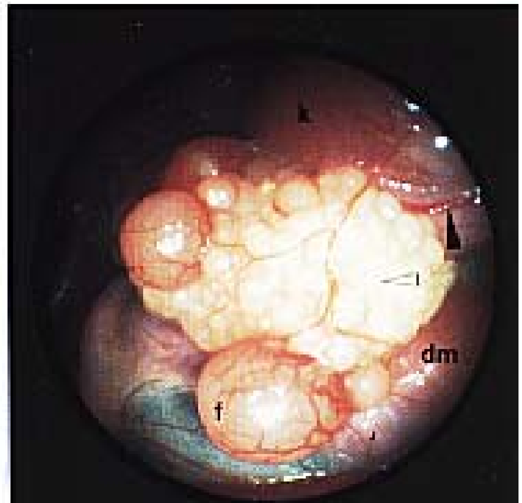
13.12



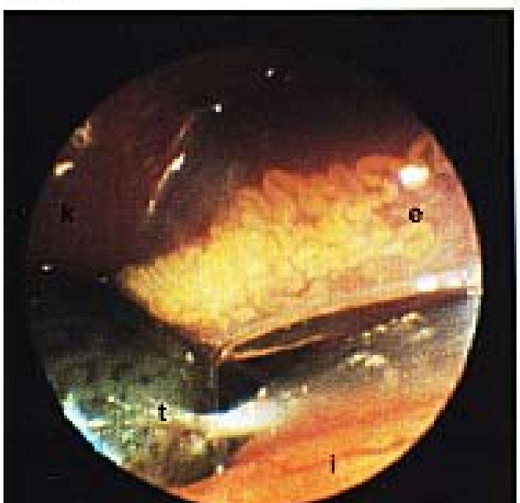
13.13



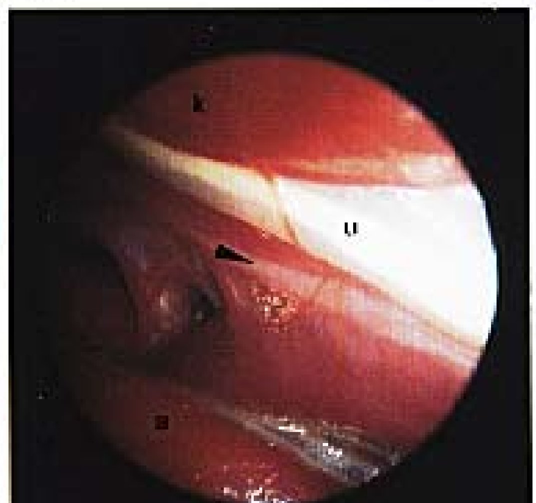
13.14



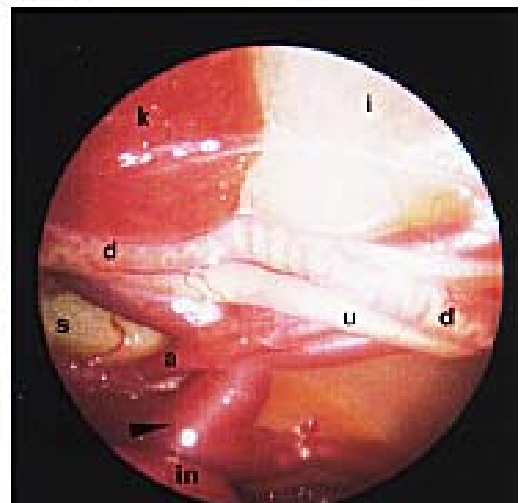
13.15



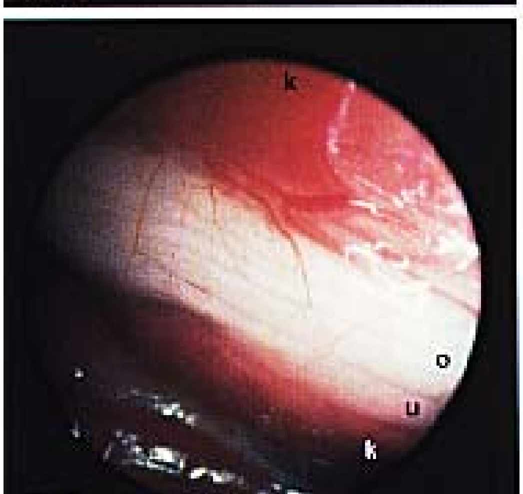
13.16



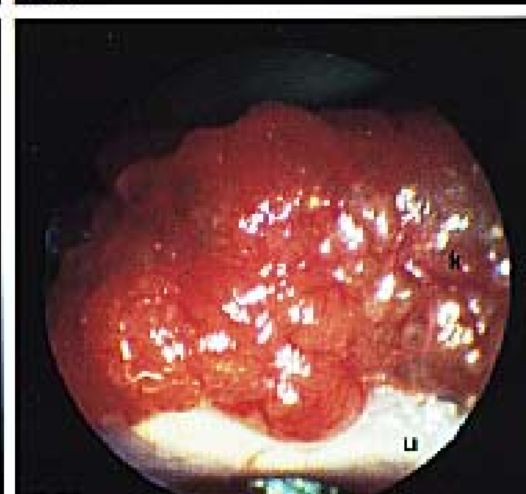
13.17



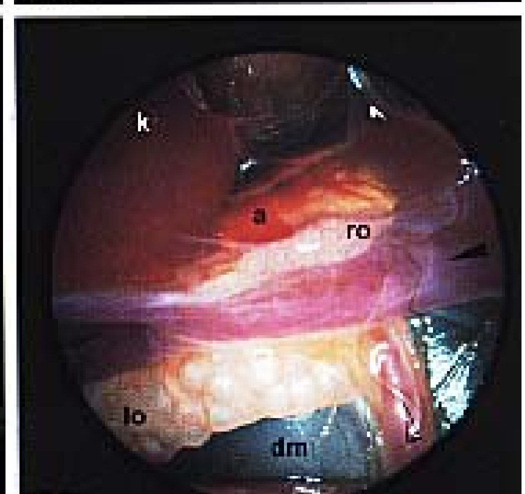
13.18



13.19



13.20



13.21

Endoscopic Examination and Biopsy Techniques

Color 13.10
(Position B-4 see Figure 13.4) Unpigmented, mature testicle (lt) in an Amazon parrot. Also noted are the lung (lu), cranial pole of the left kidney (k), epididymis (e), right testicle (rt), caudal vena cava (arrow) and right kidney (rk).

Color 13.11
(Position B-4 see Figure 13.4) Normal immature ovary (o) in a 14-week-old Amazon parrot. Also visible are the left adrenal gland (a), cranial pole of the left kidney (k), lung (lu), dorsal ligament of the oviduct (arrow) and common iliac vein (open arrow).

Color 13.12
(Position B-4 see Figure 13.4) Developing ovary (o), cranial pole of the left kidney (k), lung (lu), adrenal gland (a), dorsal ligament of the oviduct (arrow) and oviduct (open arrow). The vessels coursing across the oviduct, kidney and ovary are present in the abdominal air sac.

Color 13.13
(Position B-5 see Figure 13.4) Normal ovary in a 14-week-old Blue and Gold Macaw. The nondescript, fatty-appearing ovary (o) is difficult to identify, but the dorsal ligament of the oviduct (arrow) coursing across the kidney (k) confirms that this is a female. The vessels coursing across the kidney and ovary are in the peritoneal membrane and are seen through the abdominal air sac.

Color 13.14
(Position B-4 see Figure 13.4) Normal ovary of a mature cockatoo. The ovary (o) is melanistic and the developing follicles are translucent (arrow). The cranial pole of the left kidney (k), lung (lu), left common iliac vein (open arrow) and aorta (a) are also visible.

Color 13.15
(Position B-4 see Figure 13.4) Mature ovary of an Amazon parrot. Note the developing follicles (f) and the characteristic yellowish ("cooked egg") appearance of the involuted ovary, indicating previous ovulation sites (open arrow). The cranial pole of the left kidney (k) and dorsal mesentery (dm) overlying the right kidney are also noted. The cranial oviductal artery (arrow) is easily visualized. The vessels seen crossing the ovary are those that are present in the peritoneal membrane and are visible through the abdominal air sac.

Color 13.16
(Position C-6 see Figure 13.4) Normal epididymis (e) of an Indian Hill Mynah. Also visible are the kidney (k), caudal pole of the testicle (t) and a loop of intestines (i).

Color 13.17
(Position C-7 see Figure 13.4) Ductus deferens (arrow) of an immature macaw. Note that the ductus deferens is smaller than the ureter (u). The kidney (k) and aorta (a) are also visible.

Color 13.18
(Position C-8 see Figure 13.4) Ductus deferens (d) of a mature Amazon parrot. Also visible are the ureter (u), kidney (k), renal portal vein (arrow), synsacrum (s), ischium (i), aorta (a) and a loop of intestines (in).

Color 13.19
(Position C-7 see Figure 13.4) Oviduct (o) in a juvenile macaw. The ureter (u), kidney (k), and vessels in the abdominal air sac are also visible.

Color 13.20
Endoscopic appearance of chronic nephrosis and tubular dilation in a toucan. The abnormal kidney (k) and ureter (u) are clearly visible.

Color 13.21
(Insertion point 5 see Figure 13.2) A right abdominal (as opposed to the normal left abdominal) approach has been used to demonstrate the regression of the right ovary (ro) as the left ovary (lo) matures in an Orange-winged Amazon Parrot. Also visible are the cranial pole of the right kidney (k), the right adrenal gland (a), the caudal vena cava (arrow), the cranial mesenteric artery (open arrow) and the dorsal mesentery (dm).

Equipment

Rigid Endoscope

- ***Diameter Size:*** Fine-diameter, rod-lens endoscopes are the most suitable for avian work because of their small size, excellent optical resolution and superior light transmission capabilities. For diagnostic purposes, a 1.9 mm is the smallest diameter endoscope available with high quality optics. This endoscope is excellent for patients weighing less than 100 grams or in small anatomic sites (eg, sinus, trachea, oviduct). The major disadvantages of these very small endoscopes are their fragility, relatively small field of view and transmission of less light, which limit usefulness in larger body cavities. Because the 2.7 mm endoscope provides good light transmission capabilities with an adequate image size at a diameter that may be used in a wide range of birds, it is a good choice as the sole or principal endoscope in an avian practice[a] (Table 13.1).

  The 2.7 mm endoscope has been used in patients weighing from 55 grams to 4.0 kilograms. Intermediate-sized telescopes (eg, 2.2 mm) are available and may be preferred by some clinicians. Endoscopes (4.0 or 5.0 mm) can be employed in larger patients or when documentation demands. The advantages of the larger optics are greater light transmission and a bigger image circle. Most modern 4.0 or 5.0 mm endoscopes also incorporate new distal lens designs that provide a wider field of view; these are currently unavailable in telescopes less than 4.0 mm diameter. The author has used a 4.0 mm endoscope with wide-angle optics in birds as diverse as Golden Eagles, Crowned Cranes and Marabou Storks.

- ***Length:*** For general avian endoscopy, a length of the endoscope in the range of 170 to 190 mm is recommended. Shorter working lengths may give a more comfortable feel in use but often lack the reach desired for use in the trachea, esophagus or larger body cavities. An excessively long scope is more prone to bending or breaking.

- ***Angle of View:*** The final consideration when selecting an endoscope for avian diagnostics is the angle of view of the distal lens element. A 0° lens offset affords straight ahead viewing with a natural orientation. A 30° offset angles the field of view obliquely in the direction of the offset (Figure 13.5). This allows for improved viewing in confined areas, especially when the telescope is rotated. The bevelled distal lens element necessary to achieve this viewing angle enables easier and less traumatic passage through air sac and peritoneal walls. For these reasons endoscopes with a 30° offset are recommended for general diagnostic purposes. Specialized telescopes (eg, 70°, 90°, 130° angles) are not useful for general avian applications.

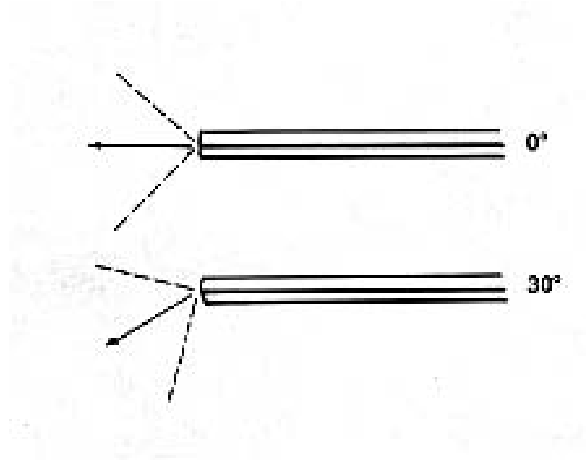


FIG 13.5 Endoscope lens with a 30° offset allows for improved vision in confined spaces.

In the late 1970's, a laparoscopic technique was devised using a veterinary otoscope as the optical device.[12] Although this instrument had the advantage of relatively low cost, it soon became clear that it could not be compared to a rod-lens endoscope in either optical quality or size of the incision necessary

TABLE 13.1 Instrumentation for Avian Endoscopy

A. Diagnostic Examination
- 2.7 mm 30° view endoscope
- Glass fiber light cable
- Diagnostic light source (150 W)

B. Minimum Diagnostic Working Set for Examination and Biopsy
- Elements listed in "A" as well as:
- Diagnostic sheath for 2.7 mm endoscope incorporating a single 5 Fr instrument channel
- 5 Fr double spoon flexible biospy forceps (oval jaws)
- 5 Fr flexible grasping forceps

C. Expanded Capabilities
- Elements listed in"A" and "B" as well as:
- Diagnostic sheath incoporating a single 7 Fr instrument channel (for larger birds)
- 7 Fr double spoon flexible biopsy forceps (oval jaws)
- 5 Fr double spoon flexible biopsy forceps (round jaws)
- 3 Fr flexible grasping forceps
- 150 W Xenon high intensity light source
- Endovideo camera

D. Other Optics
- 1.9 mm endoscope, many different lengths are available
- 2.2 mm endoscope
- 4.0 mm endoscope, 0° to 30° viewing, excellent for photo-documentation or use in larger birds

to perform laparoscopy. In the 1980's a tubular endoscope that attached to a handle-mount battery pack was introduced to the veterinary market as a less-expensive alternative to rod-lens endoscopes.[b] While this device had the advantages of lower cost, a focusing ocular and a length similar to a rod-lens endoscope, it had the disadvantages of poorer resolution, reduced light transmission and a limited field of view. The cost of a rod-lens endoscope system may be up to five times greater than less expensive instruments; however, the high optical quality, light transmission and field of view provide better long-term value when considered over the life of the endoscope. Before purchasing any endoscopic system the veterinarian is well advised to become familiar with the optical qualities of all systems under consideration. An endoscope must allow the clinician to examine tissues with accuracy and to recognize pathology or it is of no value. High quality optical systems are required to enable the clinician to achieve reliable, reproducible results. With appropriate care, modern rigid endoscopes should have a working life of five to ten years.

Veterinarians who see so few cases that they cannot justify the purchase of the appropriate equipment should refer endoscopy services to more experienced practitioners. Over the past decade, rod-lens endoscopes have become the standard for use in avian endoscopy.[2,8,21,23,27] The interests of clients and patients are best served by the use of quality optical equipment.

Flexible Endoscopes

Conventional flexible endoscopes are based entirely on fiberoptic systems for both illumination and imaging. Unlike modern rigid endoscopes, which employ solid rod-lenses, flexible endoscopes use many coherent, flexible, glass fiber bundles to transmit the image.[10] Rigid telescopes, particularly those with a small diameter, offer far better image resolution, illumination and quality than is technically possible to achieve with a flexible system. However, flexible endoscopes do provide a controllable distal tip, which allows manipulation that is not possible with a rigid rod-lens endoscope. They are most useful in examining tubular organs that are sinuous or folded. A 10 mm flexible colonoscope was found to be effective in removing lead shot from the proventriculus of Trumpeter Swans.[3] Fine-diameter, flexible endoscopes may have limited usefulness in smaller birds (eg, less than 800 g body weight) when compared to newer rigid systems.

The major disadvantage of a small-sized, flexible endoscope is that one cannot control the tip direction unless the instrument is located in a confined area such as the gastrointestinal tract. In an open area (such as the air sac), the scope cannot be manipulated or used to penetrate beyond the air sac walls without a probe. A specialty avian practice may have a small diameter flexible endoscope available to perform indicated procedures. Large flexible scopes with an operating channel for placement of grasping and biopsy instrumentation can be used in ratites.

Instrument Care

Flexible and rigid endoscopes are expensive, precision, optical instruments that will give excellent long-term performance if properly maintained. Rigid telescopes, especially those of small diameter, are fragile and must be carefully handled during transport and cleaning to avoid damage to the rod-lens elements. Torsional stresses upon the long axis of the endoscope must be avoided. This is most important when a fine-diameter telescope is being used without a protective sheath, as is frequently the case for diagnostic purposes. It is particularly important that the operator be sensitive to the amount of force being applied to the telescope during a procedure. Rigid endoscopes should always be picked up by the ocular (eyepiece) rather than the distal tip. One should lay the instrument flat to avoid bending the optical tip and fracturing the optic bundles. It is wise to clean the instrument immediately after a procedure is finished. A nonabrasive cleanser may be used to remove fat and debris. In many cases, simply washing the telescope in distilled water is all that is needed. A quality lens paper is used to clean the lens surfaces. An alcohol flush chemically dries the endoscope before it is placed in a padded storage container that

CLINICAL APPLICATIONS

- Rigid endoscopes should always be picked up by the ocular (eyepiece) rather than the distal tip.
- For office or field sterilization, sensitive endoscopic equipment may be soaked in a two percent solution of glutaraldehyde (of a type approved by the manufacturer of the equipment).
- Moderate to marked obesity leading to the intra-abdominal deposition of fat is the most frequent cause of difficulty in endoscopic visualization.
- Familiarity with anatomy, use of gentle tissue handling techniques and careful movements of the endoscope will reduce the risk of iatrogenic trauma.

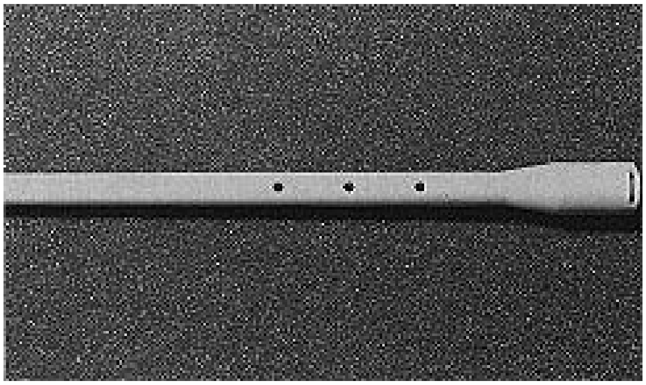


FIG 13.6 A sleeve should be placed over the endoscope for protection during movement or sterilization.

meets the manufacturer's recommendations. A simple but effective plastic endoscope sleeve is available to cover the shaft of the telescope for protection during transport and disinfection procedures (Figure 13.6).

Flexible endoscopes should also be handled with care. They should not be coiled tightly or have objects of any weight placed on the shaft, or the glass fiber bundles will be damaged. Instrument channels should be flushed thoroughly with warm soapy water to remove debris after use. Most manufacturers recommend that flexible endoscopes be stored suspended from the ocular end with the flexible shaft allowed to hang vertically. Detailed instructions for endoscope care are provided by most manufacturers. Technical staff should be properly trained in the handling and cleaning of these sensitive instruments before receiving the responsibility for their care.

Sterilization

Most endoscopic procedures require properly sterilized equipment. Even in the examination of noncritical areas such as the oral cavity or ear canal, it is prudent to remember that many animals (particularly carnivores) may harbor pathogenic organisms that can be physically transferred to another patient (particularly birds) if instrumentation is not disinfected between examinations. Due to the sensitivity of the rod-lens systems, sterilization by autoclaving is seldom recommended by the manufacturer. Expansion and contraction caused by the marked temperature extremes of steam autoclaving will damage or severely shorten the life of most telescopes. Some types of recently produced rigid endoscopes are steam autoclavable, although this process may also decrease their working life.

Two options provide safe yet consistently reliable sterilization for sensitive telescopes and light cables. Ethylene oxide gas is an extremely effective sterilant, but exposed materials must be aerated for a minimum of eight to twelve hours before use. Ethylene oxide is a human health hazard and must be used under carefully controlled conditions.

The most practical and safe alternative for the avian practitioner for office or field sterilization of sensitive endoscopic equipment is soaking in a two percent solution of glutaraldehyde (of a type approved by the manufacturer of your equipment).[c] The solution must be used according to the supplier's directions for soaking telescopes, hand instruments and light cables. The practitioner should be aware of the activated life of the product (usually 14 to 28 days) and change solutions accordingly. Stacking or layering instruments in the soak tray should be avoided so that the solution can properly reach all surfaces. Circulating the solution using a syringe is useful to ensure that all surfaces have been contacted. Minimum recommended soaking times in properly prepared glutaraldehyde solutions typically range from 15 to 20 minutes. Although greater germicidal effect is achieved the longer the equipment is soaked, many manufacturers caution against soaking for longer than two hours, as damage to glass fibers may occur.

After the soaking cycle has been completed, the equipment must be thoroughly rinsed in sterile water to prevent tissue-damaging glutaraldehyde from contacting the patient. Glutaraldehyde is extremely irritating to most tissues and may cause local irritation, tissue death, delayed healing and peritoneal reaction. Rinsing the equipment in a sterile container of sterile water for three to five minutes is most effective. The instruments are drained, immersed in a second container of sterile water for three to five minutes and wiped dry. A final alcohol wash may be used to chemically dry the equipment.

Other types of disinfectant solutions such as quaternary ammonium compounds, chlorhexidine and povidone iodine are not acceptable alternatives to two per cent glutaraldehyde solutions for soaking endoscopic equipment. With the number of resistant viruses and bacteria seen in many avian species, it is important for the endoscopist to ensure that only effective, approved products are used, or the result may be the unnecessary spread of disease.

Clinical Applications of Endoscopy

Pre-endoscopy Considerations

Indications

Endoscopic examination is indicated whenever the visual inspection of an organ or site may yield additional diagnostic information. Diagnostic endoscopy is usually preceded by less-invasive examinations such as a complete blood count, biochemistries or radiology. The patient's history, findings of the physical examination and the results of laboratory and radiologic studies may not be conclusive or may suggest endoscopic followup for additional diagnostic information (Table 13.2).

TABLE 13.2 Common Indications for Endoscopic Examination

- Loss or change in character of voice
- Acute or chronic dyspnea
- Acute or chronic sneezing
- Ingluvitis, crop burns or trauma
- Abnormal radiographic findings (plain or contrast); eg, lung, gastrointestinal tract, air sacs, organomegaly, granuloma
- Abnormal biochemical studies; eg, kidney (uricemia) or liver (elevated bile acids or liver enzyme activities)
- Persistent leukocytosis (nonresponsive to treatment)
- Acute or chronic systemic disease
- Reproductive system (suspected infertility)
- Polyuria, polydipsia
- Follow-up examination to check on lesion resolution ("second look")

- ***Diagnostic Uses:*** The endoscope and its light cable may be used to aid the physical examination.[8] The light cable may be used singly to offer additional illumination, to transilluminate a structure such as the trachea, sinus or crop, to augment examination of the oral cavity or for back lighting of overexposed radiographs. Fine-diameter endoscopes can be used in a variety of external sites where the properties of magnification, illumination and small optic diameter enhance diagnostic visualization. Many structures of the eye, ear canal, nares, oral cavity and upper respiratory tract may be examined without anesthesia. More thorough, noninvasive examinations of other body orifices are best completed under general anesthesia. The high quality optics of modern endoscopes allow visualization and inspection of tissues under magnification and are particularly useful in confined areas. Fine-diameter endoscopes introduced through a small incision, often referred to as laparoscopy,[2] permit excellent visualization of the coelomic cavities and air sacs, while creating minimal trauma.

Endoscopy has been compared to performing a necropsy on a live bird.[21] The endoscopist must become familiar with the normal and pathologic appearance of the tissues to be examined. Lesions should be described accurately regarding the location, color, size, shape and consistency. Photo or video documentation can be a tremendous aid in this process. In one study, the ability to review video recordings of examinations was believed to be an essential tool in understanding certain anatomic relationships in juvenile macaws.[29]

Improved instrumentation enhances the routine collection of specimens of suspect or abnormal-appearing tissue and debris for histologic, cytologic and microbiologic examination. Previous techniques for biopsy and specimen collection have relied on the manipulation of secondary instrumentation (eg, rigid biopsy forceps or cannulas for micro-swabs) separate from but in coordination with the endoscope. These techniques are awkward and can lead to iatrogenic trauma.[18,19,20] A new diagnostic endoscopy system for birds has recently been developed that greatly simplifies sample collection.[11,31] The system incorporates a 2.7 mm, 30° view endoscope with a single instrument port in a special sheath (Figure 13.7). Various flexible instruments may be introduced into the sheath, passed alongside the endoscope and guided to a specific site with great ease (Figure 13.8). Iatrogenic tissue trauma is markedly reduced because the instruments are directed to the visual field through the integral sheath, avoiding the blind manipulation required to place a second, rigid instrument.

- ***Surgery:*** Harrison[8] first suggested the use of the endoscope as an operating telescope in open avian surgery to enhance visualization of small structures.

Endoscopic surgery is currently one of the fastest growing areas in the human surgical specialties. Special hand instruments have been developed to enable tissue manipulation, suture and clip placement and radiosurgical techniques using the endoscope. The advantage of this type of surgery in humans has decreased patient trauma and hospitalization. The technology offers great promise if it can be adapted for avian surgery. In addition to decreased trauma, the magnification and illumination provided by a quality endoscopic system enable more precise techniques in small avian patients. Surgical procedures

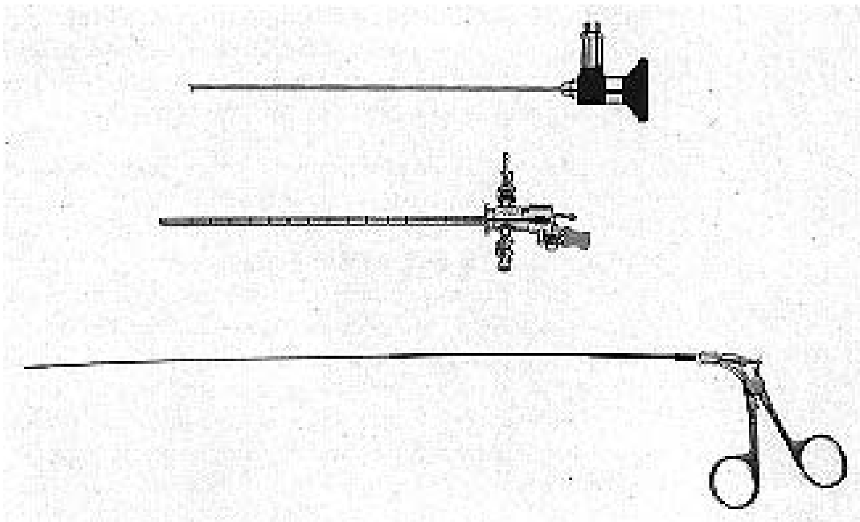


FIG 13.7 A specialized 2.7 mm endoscope that fits into a sleeve is ideal for most avian endoscopic procedures. The sleeve has been designed to accomodate the introduction of biopsy forceps and other flexible instruments to facilitate the collection of diagnostic samples (courtesy of Karl Storz Veterinary Endoscopy–America).

are under development for endoscope-guided hysterectomy, ulcer repair and egg removal.

- ***Identification of Gender of Monomorphic Birds:*** The use of endoscopy to identify the gender of monomorphic birds was the earliest widespread application of this technology in avian medicine; it has been the major force supporting the development and introduction of improved diagnostic capabilities utilizing endoscopy.

Contraindications

The general contraindications for endoscopy are those that would apply to general avian surgery and anesthesia. Moderate-to-marked obesity leading to the intra-abdominal deposition of fat is the most frequent cause of difficulty in endoscopic visualization. Large peritoneal fat reserves may make the examination of parts of the coelom impossible. In some cases, an improved diet is recommended for the patient (with reexamination in six to eight weeks).

The presence of ascites may cause difficulties if the peritoneum of the ventral hepatic peritoneal cavity (VHPC) or intestinal peritoneal cavity (IPC) is breached while entering the air sac. Fluid could drain from the peritoneal cavity into the air sac and from there into the lung, leading to aspiration and death. This is most likely to happen in a lateral approach to the caudal thoracic air sac. If ascites is suspected and an endoscopic examination of the liver is necessary, the ventral approach to the VHPC should be used. Fluid from the VHPC will drain from the incision site and can be safely suctioned without the concern for air sac involvement.

Left coelomic examinations should not be performed in the hen near the time of ovulation, as the ova greatly enlarges in size, virtually obliterating the abdominal air sac. The oviduct also increases in size and tortuosity, filling the left portion of the IPC. Use of the post-pubic approach to the abdominal air sac risks damage to the oviduct or an egg nearing oviposition. A left lateral coelomic approach is rendered less useful by the presence of large, developing ova that makes visualization difficult. Attempting passage into the abdominal air sac from the caudal thoracic air sac may be difficult and potentially risks damage to the ova.

Inexperience of the operator remains one of the most common causes of endoscopic complications.[8,21] Veterinarians considering the addition of endoscopic services to their avian practice are well advised to seek out appropriate continuing education and to become thoroughly familiar with normal avian anatomy. Experienced colleagues may be contacted for advice on practical equipment needs before purchasing new equipment. Necropsy specimens can be used to study endoscopic principles and are particularly useful in learning to identify tissue changes. Lectures and laboratories are available on endoscopic techniques.

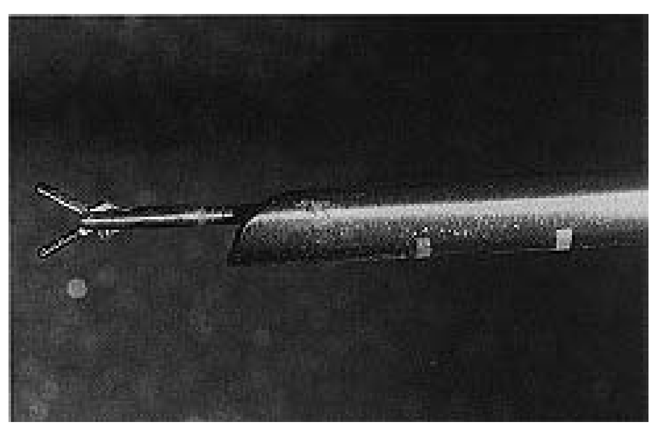


FIG 13.8 Biopsy forceps passing through a specially designed sheath for a 2.7 mm endoscope (courtesy of Karl Storz Veterinary Endoscopy–America).

Complications

As part of the informed consent process, the client must be made aware of potential complications of the endoscopic process. Anesthesia-related incidents are described in Chapter 39. Organ trauma is one of the most serious intraoperative endoscopic complications. The proventriculus may be punctured using a trocar and cannula or similar entry device from a lateral approach. Failure to identify and repair this injury can result in a fatal peritonitis. Laceration of a blood vessel or organ such as the liver or spleen is possible and may lead to serious or fatal hemorrhage. Liver or kidney contusions can be caused by the endoscope tip during excessively vigorous manipulation. These are infrequently the cause of serious clinical problems.

Familiarity with anatomy, use of gentle tissue handling techniques and careful movements of the endoscope will reduce the risk of iatrogenic trauma.

Subcutaneous emphysema is a potential (if rare) postoperative complication. Air may leak through the body wall at the point of the air sac entry and collect under the skin. Usually, the body wall opening will seal without incident, but occasionally the skin incision must be reopened and the body wall puncture sutured. This is most common when a large endoscope or sheath has been used. Endoscopic punctures may be routinely closed with a fine-diameter, absorbable, monofilament suture.

Air sac and peritoneal granulomas can occur by using instrumentation that has been improperly sterilized or in situations where poor technique or inadequate skin preparation has allowed contamination of the endoscope tip. The peritoneum of most birds seems to be quite forgiving of small insults. Many granulomas may not be appreciated clinically. The author and editors have never experienced a case of generalized sepsis related to endoscopic manipulation, although this may be possible in cases where the wall of an existing granuloma is damaged or where ineffective sterilization practices have been used. Cold disinfectant solutions such as chlorhexidine[2] are not appropriate, as some organisms such as *Pseudomonas aeruginosa* will survive this treatment. No comprehensive studies have examined the endoscopy and biopsy sites days to weeks following the procedure. However, preliminary work indicates that there was no local infection in patients where a two percent glutaraldehyde solution was used for instrument preparation and appropriate sterile technique was employed. The inability to perform proper sterilization of a single endoscope makes surgical sexing clinics obsolete. Transmission of viral infections (particularly Pacheco's disease virus and polyomavirus), resulting in the loss of numerous birds has been linked to "sexing clinics." The mixing of birds from multiple sources (particularly when an invasive procedure is performed) should be discouraged. It is possible, however, to safely perform endoscopy on several birds from a single client by utilizing two endoscopes, with one being sterilized while the other is in use.

Patient Preparation

Patients should be fasted a minimum of three hours. In some cases the length of the fast is extended, especially if the endoscopic examination will involve the gastrointestinal tract. Species that consume large boluses of whole foods (eg, raptors) may require fasting for 24 to 36 hours. Failure to do this may make examination of many portions of the coelom impossible due to distension of the proventriculus.

Surgical sites are prepared as for any avian surgery. Particular attention should be paid to the skin surface. The endoscope can transmit surface debris into the body cavity if the entry site is not properly prepared. Small, sterile, transparent, wound dressings[d] make excellent drapes for endoscopic procedures. They are available in a variety of sizes and are lightly adhesive so they stay in place without clamps; their transparency improves anesthetic monitoring in small patients.

- ***Anesthesia:*** Appropriate anesthesia is an essential part of good endoscopic practice. It is seldom possible to perform an endoscopic examination in the physically restrained bird without the risk of organ contusion or other trauma. Consistency in positioning of the patient is mandatory for anatomic orientation. Maintaining position is neither possible nor humane using physical restraint only. Clinical anesthesia has been thoroughly reviewed in Chapter 39. The anesthetic agent of choice for most endoscopic procedures remains isoflurane.[e]

Sites of Application

Air Sacs, Lungs

Birds are excellent subjects for endoscopic examination because the unique system of air sacs allows visualization of coelomic structures without artificial insufflation. Air sacs invaginate the thoracoabdomen of birds to facilitate examination of or access to specific organs. There are marked similarities in the morphology of caudal air sacs among selected Passeriformes, Psittaciformes, Columbiformes, Gruiformes, Strigiformes and Falconiformes. In chickens, there are three paired air sacs (cranial thoracic, caudal thoracic, abdominal) and two single, median air sacs (cervical, clavicular).[17]

There is one published examination of air sac morphology in a psittacine bird (budgerigar).[4] The budgerigar has paired, unfused cervical air sacs but was otherwise similar to the chicken. The caudal thoracic air sacs of the pigeon extend farther caudally than in most Psittaciformes. In some diving birds, the caudal thoracic air sacs are much larger than in other species. This is assumed to be an adaption to increased air requirements while diving underwater.

For endoscopic purposes, it is preferable to consider the cranial and caudal thoracic and the abdominal air sac pairs together. In the parrot, the cranial thoracic air sacs are the smallest of the group and are located ventral and cranial to the caudal thoracic air sacs (Color 13.1). They are best accessed from the ventrolateral thoracic wall using the approach first described by Bush,[2] who suggested an entry site caudal to the last sternal rib in the area of the lateral notch (a "V"-shaped depression palpable between the sternum and the last rib).

The patient is placed in lateral recumbency with the wings extended dorsally. The wings may be taped to a restraint surface or they may be affixed with a short loop of non-adhesive, self-adhering tape[f] passed between the primary feathers and around the carpus. The landmarks are located and a small skin incision is made. The musculature of the body wall is bluntly separated and the endoscope is inserted in a craniodorsal direction. From this approach the pericardial sac and heart can be seen as well as the lobe of the liver and the caudal, ventromedial surface of the lung (Color 13.23).

The traditional left lateral surgical approach takes advantage of the air sac anatomy to approach the gonads by either directly entering the abdominal air sac or by entering the caudal thoracic air sac first and then passing into the abdominal air sac through a small incision (see Figure 13.2). This approach is similar to the early laparotomy techniques of field ornithologists.[1,26]

The patient is placed in true lateral recumbency with the wings extended dorsally. The upper leg is extended and held caudally. The point of insertion is located by palpating the triangle cranial to the muscle mass of the femur, ventral to the synsacrum and caudal to the last rib.[8,19,23] The body wall may be penetrated by a trocar and cannula or by blunt separation. In Psittaciformes, this entry site has been demonstrated to occur between the seventh and eighth ribs (not the behind the last rib). With this approach, the tip of the endoscope enters the mid to caudal portion of the caudal thoracic air sac in most birds.

As an alternative approach to the caudal thoracic air sac, the bird is restrained in lateral recumbency except that the leg is extended cranially.[15] The site of entry is the same as previously described in the upper part of the triangle formed by the proximal femur, the last rib and the cranial edge of the pubis.

A similar approach to the caudal thoracic air sacs that is based upon precise landmarks has been developed (see Figure 13.3).[29,30,31] The animal is positioned as described. The entry site is located by finding the point where the semimembranosus muscle (M. flexor cruris medialis) crosses the last rib (Color 13.2). The ventral fascia of the semimembranosus muscle is bluntly separated from the underlying body wall and the muscle is reflected dorsally. A blunt entry is made just caudal to the last rib, beneath the reflected semimembranosus muscle. Except in individuals with moderately to markedly increased fat reserves, the landmarks are located easily. The procedure is reproducible in members from a wide variety of orders including Psittaciformes, Passeriformes, Columbiformes, Gruiformes, Falconiformes and Strigiformes. A major advantage in placing the leg forward is that the lateral body wall can be more easily approached without the interference of the femoral musculature. This becomes particularly important in birds with heavily muscled upper thighs (eg, many Psittaciformes).

With either of these approaches the endoscope enters the caudal thoracic air sac at or near its caudal border. Color 13.4 was photographed from the left entry point of this caudal approach looking cranially. Visible from eleven to one o'clock is the caudal surface of the lung with its large ostium. From the two to three o'clock position is the transparent membrane formed by the confluent walls of the caudal thoracic air sac and the abdominal air sac. Passing through this wall would place the endoscope within the abdominal air sac. At four to six o'clock is the ventrolateral border of the proventriculus. The lateral edge of the left lobe of the liver may be seen at the seven to eight o'clock position. From nine to ten o'clock is another transparent membrane. This one is composed of the walls of the confluent caudal thoracic air sac and cranial thoracic air sacs. Passing through this membrane would place the tip of the endoscope in the cranial thoracic air sac.

The abdominal air sacs of most birds are the largest air sacs. They extend from the caudal surface of the lung to the craniolateral borders of the cloaca. Entry into the abdominal air sacs may be gained through one of the previously described caudal thoracic air sac approaches or by direct access through the caudal body wall. Lumeij[21] was the first to describe a postpubic approach to the caudal portion of the abdominal air sac. The entry point is situated dorsal to the pubic bone and caudal to the ischium (see Figure 13.2). The endoscope generally first enters the most caudal portion of the intestinal peritoneal cavity and must be penetrated through this thin membrane to enter the abdominal air sac. The endoscope can then be moved cranially up the length of the abdominal air sac. From the left approach a large number of structures may be examined including the kidney, adrenal, gonad and associated structures, spleen, proventriculus, ventriculus and intestine (Color 13.26). The abdominal air sac may also be approached from a flank position. The entry site is located directly ventral to the acetabulum and just dorsal to the ventral border of the flexor cruris medialis muscle.

Reproductive Organs

In most avian species, only the left ovary and oviduct develop.[14,16] The development of the right ovary is normally arrested in a testis-like stage and can frequently be visualized near the right adrenal gland, along the caudal vena cava (Color 13.8). For this reason, endoscopy to examine gonadal structures is performed through the left side of the abdomen.

The testicle of the adult male bird is ellipsoidal to bean-shaped. In most species it is creamy white although it may be more or less pigmented (gray to black) in others (eg, cockatoos, mynahs, toucans) (Color 13.7). Under the seasonal influence of hormones, the mass of the testicle may increase from 10 up to 500 times.[14] The pattern of surface vessels increases and becomes more prominent. The epididymis enlarges, and the ductus deferens becomes very tortuous in preparation for storage and transportation of the spermatozoa (Color 13.16).

In contrast, the ovary of the mature female has the appearance of tapioca pudding with many small follicles visible during the nonbreeding season. Under appropriate hormonal stimulation, a hierarchy of follicles develops and matures giving the ovary the appearance of a cluster of grapes (Color 13.15). A follicle enlarges as it matures; simultaneously, the oviduct increases in size and becomes tortuous and folded in preparation to accept the ovum. A large ovum can be mistaken for a testicle, especially in an obese bird where other structures are difficult to see or where the surgeon fails to check related anatomic reference points.

The differences in the morphology of adult gonads are relatively distinct. In juvenile birds, gonadal tissue is less obvious and differentiation is more difficult. It is possible to endoscopically identify the correct gender of most species of birds at a young age if good optical equipment is used and a careful examination of the gonads and associated structures is performed.

In one study of juvenile macaws,[29] differentiation of the sexes was uniformly possible as young as six weeks of age when gonadal and oviductal or ductus deferens morphology were considered together. Testicles were tubular to ellipsoidal with distinct, rounded cranial and caudal poles. A paired right testicle could usually be seen through the dorsal mesentery (Color 13.10). The ductus deferens was a thin, white tubular structure, usually only one-third the diameter of the ureter (Color 13.17).

The juvenile ovary was comma-shaped, dorsoventrally flattened and closely applied to the adrenal and cranial pole of the kidney. The surface texture of the ovary was dependent on the age of the bird. Very young ovaries had a faintly granular surface with fine sulci (Color 13.13). As the birds aged, the sulci deepened, giving the ovary a furrowed, brain-like appearance (Color 13.12). With the maturation of the primary oocytes, the ovary began to take on a distinctly granular texture with a more three-dimensional shape, and the sulci disappeared (Color 13.14). The oviduct was pale white with a thicker, more substantial appearance than the vas deferens. The oviduct was generally two to four times the thickness of the ureter and on close inspection, fine, longitudinal, spiral bands were visible (Color 13.19). These may have represented the developing spiral folds of the mature oviductal mucosa. The most interesting finding of this study was the presence of the supporting ligament of the infundibulum, which was clearly visible crossing the cranial division of the kidney. This structure is part of the dorsal ligament of the oviduct and is absent in juvenile males (Color 13.11).

From this approach, it may be difficult or impossible to view the right side in mature birds. Examination of the right abdominal air sac (AAS) would be required to confirm the presence of abnormalities related to the remnant ovary or the right testicle such as ovotestes or hermaphrodism. While these conditions are uncommon, their presence may need to be ruled out in cases of infertility.

Caution should be exercised in estimating age, reproductive history or reproductive potential based upon a single endoscopic examination. During the nonbreeding times of the year, the adult gonads return to a quiescent state similar to those of the late adolescent bird. Several male Hyacinth Macaws in their teens had very small testicles, yet went on to breed within months of evaluation. A mature African Grey Parrot showed no evidence of follicular development at examination but ovulated 24 days later.

During the endoscopic examination for gender determination, the endoscopist is able to evaluate the air sacs, liver, lung, spleen, kidney, adrenal gland, proventriculus, ventriculus and the visual portions of the intestines. A systematic examination that may suggest a subclinical health problem can provide data of value to the aviculturist. This information is not available using cytogenetic or molecular biological techniques of gender determination.

Ear Canal

The external auditory meatus is hidden by specialized covert feathers that lack barbules. There is no pinna. The opening is usually rounded but can vary in diameter from small (2.0 to 15.0 mm in passerine and psittacine birds) to very large (up to 6.0 cm in owls). The ear canal is straight and short. The tympanum can usually be visualized clearly (Color 13.41). A 1.9 mm telescope is often needed to explore the deeper aspects of the canal. Unlike the dog and cat, birds infrequently suffer from otitis externa.

Oropharynx

The oral cavity is easily approached in most avian species. The bill may be held open manually or with a speculum. In species with strong mandibular musculature (such as Psittaciformes) it is recommended that the patient be anesthetized for most oral examinations. If manual restraint is used, extra care must be taken to prevent damage to the equipment.

The avian tongue may exhibit a number of adaptations for food prehension and manipulation. In many species it is a flat, triangular-shaped organ with a relatively smooth epithelium. Psittaciformes have large, fleshy tongues ideally suited to food manipulation. They are the only order with intrinsic lingual muscles[17] that allow a great variety of movement and flexibility. In many species, including parrots (Color 13.34), there are a group of mucus-secreting salivary glands at the base of the tongue. Inspissation of keratinized debris due to squamous metaplasia will be seen in birds suffering from hypovitaminosis A (Color 13.33).

Salivary glands are most prominent in species that eat primarily a dry diet (cereal grains) and may be absent in those that eat a moist, lubricated diet (fish). In the parrot, salivary glands are found along the roof and the floor of the mouth and on the tongue. The oropharynx is lined with stratified squamous epithelium and may be keratinized in areas of wear. In some species, the epithelium may be heavily pigmented. It normally has a smooth, unblemished surface except in areas where spike-like sensory papillae are present (Color 13.34). The mucosa should be examined for adherent exudate, debris or ulcers, as may be seen in certain protozoal (eg, *Trichomonas* sp.), fungal (eg, *Candida albicans*) or viral (eg, poxvirus) diseases.

The choanal slit is visible as a median "V"-shaped cleft in the palate. There is species variation in the width of the choanal borders. In pigeons and most raptors, the choana is slit-shaped (Color 13.35). In the parrot the borders are more widely spaced, forming a distinctive "V" shape. The borders of the choanal slit are lined with sensory papillae. By entering the choanal slit with the scope and moving craniodorsally, the nasal septum and conchae can be examined (Color 13.37). Just caudal to and on the midline of the choana is the small slit-like infundibular cleft. This is the common opening of the right and left pharyngotympanic tubes[17] also referred to as the eustachian tubes (Color 13.35).

The laryngeal mound is visualized at the base of the tongue on the midline of the caudal floor of the oropharynx. The paired, fleshy laryngeal prominences open and close to form the conspicuous glottis. There is no epiglottis (Color 13.31).

Trachea

The trachea may be entered at the larynx by passing through the glottis of an anesthetized patient. The avian larynx does not contain vocal cords. Tracheal rings of the bird are usually calcified and are completely circular. The tracheal mucosa consists of smooth, stratified squamous epithelium. The syrinx is the site of sound production and is located where the trachea bifurcates into the primary bronchi. The syringeal membrane may be the site of opportunistic bacterial and fungal infection (aspergillosis).[25] Tracheoscopy to the level of the syrinx is possible in medium-to-large birds using a 180 mm long, 2.7 mm endoscope. Smaller patients (larger than cockatiels) may be examined with a l.9 mm endoscope. Visualization can be improved by extending the neck. In patients with acute to subacute dyspnea, tracheoscopy should be considered to rule out foreign objects or inflammatory debris.

In seed-eating birds, hulls or whole seeds may be aspirated into the larynx or syrinx. In carnivorous birds, small pieces of bone, tendon or cartilage may become lodged in the glottis. These may be removed using endoscopically guided grasping forceps. Tracheitis may be caused by bacterial or viral agents. Culture of the endoscope tip immediately after removal from the patient may be helpful in determining an etiologic agent.

Esophagus and Ingluvies

The esophagus is easily entered by passing the endoscope caudally into the pharynx and over the laryngeal mound. The surface of the esophagus is comprised of longitudinal folds that vary depending upon the dietary habits of the species (see Color 19). For example, the number and size of folds and the degree of distensibility are less in insectivores and seed eaters than in carnivores like hawks and owls (Color 13.38).

It is a common misconception that all birds have an ingluvies. Galliformes, Psittaciformes, Columbiformes and some Passeriformes have a true crop. The ingluvies can be examined with either a flexible or rigid endoscope after passing the instrument through the cervical portion of the esophagus. Insufflating the crop with air will help with visualization. To do this, a small-diameter, flexible feeding tube,[g] which has been attached to a 35 or 60 cc syringe, can be passed into the crop (Color 13.39). Alternately, the insufflation channel on a 4 mm or greater diameter flexible endoscope, or the instrument channel on the Storz rigid avian sheath[a] can be used to distend the ingluvies with air. Some pressure will need to be maintained around the proximal cervical esophagus to retain the infused air within the crop. Patients undergoing elective ingluvioscopy should be fasted for several hours before the procedure to reduce the effects of retained food materials upon visualization. With this technique the crop mucosa can be thoroughly examined and small foreign objects can be removed with grasping forceps. The grasping forceps can be endoscopically guided using either a flexible endoscope with an instrument channel or the rigid sheath with channel.

Proventriculus, Ventriculus

The proventriculus and the ventriculus may be examined using either flexible or rigid equipment. In a 250 to 600 g parrot it can be a difficult chore to guide a small-diameter, flexible endoscope down the cervical esophagus, across the crop and into the thoracic esophagus, although this equipment can be used successfully in larger parrots and in moderately large avian species that lack a crop (eg, owls and Anseriformes). A pediatric bronchoscope is required in smaller patients. The smallest practical flexible endoscopes with an instrument channel are pediatric bronchoscopes at 4.0 mm and 5.0 mm diameter. Human flexible colonoscopes (10.0 mm) have been shown to be useful in very large species such as swans and cranes.[3] Once the endoscope is positioned in the thoracic esophagus, the pathway becomes a relatively straight one continuing into the proventriculus (Color 13.40) and the ventriculus.

Preliminary studies using a midline ingluviotomy to enter the thoracic esophagus using the Storz 2.7 mm rigid endoscope and instrumented sheath have been performed.[a] Birds were anesthetized, intubated and placed in dorsal recumbency.

Care was taken to avoid the passage of proventricular contents into the trachea or choana by inserting an absorptive gauze tampon into the cranial cervical esophagus and ensuring that the endotracheal tube was secure. Whenever possible, patients were fasted for five to six hours in order to empty the proventriculus. In cases where fasting was not possible (eg, acute foreign body ingestion), the proventriculus was flushed with sterile saline and ingesta was forced out of the thoracic esophagus and into the crop, from which it was suctioned. Placing the patient with its head down facilitated this procedure. A small skin incision was made over the middorsal portion of the crop. The crop wall was incised. The entrance to the thoracic esophagus was located on the ventral midline border of the crop, and the telescope and sheath were introduced.

The sheath and endoscope were inserted into the thoracic esophagus. A 3-5 Fr rubber catheter connected to a syringe containing saline was inserted into the instrument channel for use in flushing debris from the visual field. Grasping forceps[j] (3 Fr or 5 Fr) can be inserted into the channel to manipulate and remove foreign objects. The crop incision was closed using standard techniques.

Ventral Hepatic Peritoneal Cavities

The liver of the bird is encapsulated within two paired peritoneal cavities: the ventral and dorsal hepatic peritoneal cavities. The paired ventral hepatic peritoneal cavities (VHPC) are the largest and of greatest clinical significance. The right and left VHPC are separated by the ventral mesentery. The right lobe of the liver is larger in most birds (Colors 13.22, 13.28).

To gain access to the liver, one or both of these ventral cavities must be entered. The liver can be visualized from the cranial and caudal thoracic air sacs (Colors 13.5, 13.24) and indeed seems tantalizingly close in most birds. In reality, the liver is covered by a layer of peritoneum that is contiguous with the overlying air sac. To access the liver, the ventral hepatic peritoneal cavity (VHPC) must be entered either laterally from the caudal thoracic air sac or by a direct, ventral midline approach. The ventral approach[16] is best for examining and sampling both lobes of the liver. A skin incision is made on the midline just caudal to the border of the sternum. The linea alba is incised and the caudal border of the VHPC is bluntly penetrated. A substantial fat pad may be present overlying the outer surface of the caudal border of the VHPC. Under conditions of health, the liver should not protrude past the caudal border of the sternum.

The VHPC may also be entered from the caudal thoracic air sac. This may be most convenient when a lateral approach has been used for a general diagnostic examination and liver lesions have been noted. An opening can be made in the confluent walls of the caudal thoracic air sac and the VHPC by using endoscopically guided forceps to pick up and tear a small hole in the membranes. The lateral border of the liver can then be grasped through this VHPC access (Colors 13.29, 13.42). This approach is contraindicated in patients with ascites because fluid will drain into the air sac and may be aspirated (Color 13.30).

Intercostal Approach to Lungs

An intercostal approach to the lung for biopsy has been recently described in the pigeon.[11] Entry was recommended through the dorsolateral portion of the third or fourth intercostal space where pulmonary tissue is the thickest in cross section. The third intercostal space is located by counting cranially from the last rib. The space is palpated just ventral to the scapula and a small skin incision is made. The intercostal muscles are bluntly separated to the level of the pleura. Care must be taken during dissection through the intercostal muscle to avoid deep penetration, which can traumatize the surface of the lung. The resulting hemorrhage may make visualization difficult and lead to sample artifact.

An instrumented sheath and rigid endoscope are inserted into the incision and maneuvered carefully between the ribs so that the surface of the lung can be visualized. The rounded edges of the sheath aid in atraumatically positioning the instrument. A 5 Fr flexible forceps is advanced into the lung parenchyma, the jaws closed rapidly and removed. Post-biopsy hemorrhage may vary from mild to moderate but is usually controlled by pressure. Intercostal muscle and skin are closed routinely with simple interrupted sutures.

While it is not essential to utilize an endoscope to biopsy the lung from this site, it was found that the sheath and endoscope combination greatly aided the collection of quality pulmonary biopsies with less risk of trauma to the patient.[11] Rigid cup biopsy forceps can be manipulated unaided through a similar intercostal incision, but trauma to the surface of the lung may be greater due to the short working distance and lack of magnification.

Intestinal Peritoneal Cavity

The intestinal peritoneal cavity (IPC) is the largest of the peritoneal cavities. It is a single, midline potential space that extends from the level of the kidneys caudal to the vent. It is somewhat subdivided by the several mesenteries formed by reflections of the peritoneum that suspend the proventriculus, intestines, gonads and supporting structures.[16,24] The gonads are actually suspended within the IPC and are not located within the abdominal air sac. The confusion in this positioning is understandable because the gonads are clearly visible from the abdominal air sac even though they are covered by the air sac wall and the confluent peritoneum (Color 13.12, 13.13). One method to demonstrate the relationship between the IPC and the abdominal air sac is to insert an endoscope into the IPC, optically guiding it toward the left gonad and then viewing this arrangement from the abdominal air sac via a second endoscope (Color 13.25). The thin but substantial air sac/peritoneal wall can be seen clearly covering the endoscope.

The intimate relationship between the abdominal air sac and the IPC is of greatest clinical significance in the female bird. The dorsal mesentery, the dorsal parietal peritoneum and the peritoneum covering the left abdominal air sac fuse to form a serous pocket surrounding the ovary.[24] This "ovarian pocket" is believed to help guide ova to the infundibulum.[6] It has been suggested[5] that extensive damage to both the right and left abdominal air sacs in female birds will lead to infertility and that trauma should be limited to only one of the abdominal air sacs. This suggestion ignores the presence of the IPC and simplifies the role of the abdominal air sacs. Under routine endoscopic examination from a lateral approach only the lateral wall of the abdominal air sac is penetrated. The confluent medial wall of the abdominal air sac and the IPC would not be penetrated under these circumstances. Thus, the ovarian pocket would not be disrupted. A hysterectomy (salpingohysterectomy) performed from a lateral approach will disrupt the left IPC membrane.

Cloaca

The cloaca is a unique, three-chambered structure that receives the terminal portions of the colon, ureters and reproductive tract. Endoscopic examination of the three parts of the cloaca is complicated by the presence of feces and urates. Flushing the proctodeum with saline and then insufflating the structure while closing the vent lips around the telescope will enhance viewing. Uroliths,[18] papillomatous inflammation and true prolapse have been documented with endoscopy. Bacterial and fungal cloacitis may also occur.

Distal Oviduct (Uterus)

Endoscopic examination of the distal oviduct (uterus) is possible in reproductively active birds and may be a useful procedure for the sampling and diagnosis of oviductal disease.

Biopsy Techniques

Patient Considerations

Indications

Open (surgical) and percutaneous techniques for biopsy of the liver have been described in avian medicine. Other internal organs have occasionally been biopsied using open techniques. The ability to obtain precise target biopsies of specific organs is a natural extension of endoscopic examination and offers a far less traumatic method for obtaining diagnostic specimens. Carefully selected biopsies of affected organs may be critical in establishing a diagnosis and allow more precise therapeutic decisions. Table 13.3 describes approaches and techniques for specific organ biopsies.

Indications for biopsy may include abnormal radiographic findings or biochemical parameters, chronic respiratory disease, polyuria and polydipsia (see Table 13.2). The endoscopist should be prepared to collect biopsies during routine examinations. It is not uncommon to find unexpected lesions in patients presented for gender determination. Specimens from obvious lesions are easily collected from the border zone where abnormal meets normal tissue. If the patient's history, physical examination or biochemical findings suggest renal or hepatic abnormalities, biopsy of the kidney or liver is indicated. Tissues will frequently appear grossly normal even though there are significant histologic lesions present.[20]

The decision to biopsy the liver or kidney (Color 13.43) is frequently made too late in the disease process to be truly helpful to the patient and client. Sampling the end stage liver is seldom illuminating beyond confirming a poor prognosis that should be otherwise clinically evident. Many birds with early cases of hepatic disease demonstrate few clinical signs. Recent advances in avian clinical biochemistry procedures, particularly the measurement of bile acids, promise to improve the clinician's ability to detect liver disease at an early stage. Bile acids determination is a sensitive and specific indicator of liver damage (see Chapters 11, 20). Histologic changes are seen in the livers of patients with persistent increases in the bile acids of two times or greater the normal reference intervals. A liver biopsy is recommended in cases where the bile acids measurement remains elevated following the completion of therapy for a systemic disease (eg, chlamydiosis) or where continued elevation of two weeks or more is confirmed.

Renal disease can also be challenging to recognize and diagnose in its early stages (Color 13.47, 13.48). Elevations in uric acid levels may not occur until a relatively large number of renal tubules have been damaged. Polyuria is frequently noted. Kidney biopsy is recommended when uric acid levels are consistently above reference values or show evidence of

TABLE 13.3 Specific Organ Biopsies: Approaches and Techniques

Organ	Approach	Technique
Liver	Best accessed from the VHPC but may also be approached through the left and right caudal thoracic air sac (caudal TAS).	In generalized diseases of the liver, samples can be most easily obtained from the hepatic border using a 5 or 7 Fr instrument. In focal disease (eg, granulomas, neoplasia), the lesion should be specifically targeted taking care not to open the jaws of the forceps too wide when pushing into the liver. This will reduce crush artifact. Larger, rigid forceps can be used but are not usually necessary.
Kidney	Through the caudal TAS into the cranial portion of the abdominal air sac (AAS) or via the AAS approach. May also be approached directly through the IPC, although this potential space would need to be insufflated. The caudal TAS approaches are most suitable for reaching the cranial and middle divisions of the kidney. Entry into the AAS is an excellent way to reach the caudal division of the kidney.	Depending upon the size of the patient, 3, 5 or 7 Fr forceps can be used. Cup-shaped forceps can be used to control depth of penetration. In some smaller birds, the 5 Fr round cup forceps[j] may be more appropriate than those with a standard elliptical shape.
Air Sac	In most species, the caudal TAS is the one most frequently involved in air sac pathology. Lesions may be more prominent on one side than another or may involve the cranial TAS more extensively. Radiographs may be most helpful in selecting the preferred entry site.	Cup biopsy forceps may be used to grasp a small piece of air sac from the border of an air sac puncture site (eg, the caudal TAS/AAS entry site) or to harvest focal lesions directly from the surface of the air sac. Exudate may also be collected with the forceps for microbiology.
Lung	Two approaches to the avian lung were recently described.[11] An endoscopically-guided biopsy of the caudal surface of the lung can be collected from the caudal TAS using the Storz system. It is also possible to access the costal surface of the lung through an intercostal approach. The Storz system may be used to enter the intercostal space and visualize the lung, or a rigid forceps may be guided by the surgeon unaided.	5, 7 and 9 Fr forceps have been used to collect pulmonary biopsies. The degree of localized pulmonary hemorrhage is directly related to the size of the biopsy forceps used and the depth of penetration.
Spleen	The spleen is approached from the left AAS. The AAS may be entered through the caudal TAS or from the caudal approach. The spleen is located on the right side of the proventriculus near the junction with the ventriculus.	A 5 Fr elliptical cup is satisfactory for most patients.
Ventriculus	The greater curvature of the ventriculus, particularly the caudoventral surface, is best approached from the caudal TAS through the left paralumbar fossa.	A 7 or 9 Fr elliptical forceps is recommended. A minimum of two biopsies is collected from the caudoventral surface near a blood vessel to ensure the harvesting of nerve as well as serosa and muscularis.
Testes	The right or left testicle can be reached from its respective AAS or from the IPC.	A smaller forceps is less traumatic (eg, 5 Fr round) although testicular biopsy utilizing a 9 Fr elliptical cup instrument has been reported.[8]

an increasing trend or where polydipsia and polyuria persist without clinical explanation.

Air sac and pulmonary biopsies are indicated when clinical examination, radiographic studies or auscultation reveal persistent, nonresponsive respiratory disease. Dyspnea upon exertion is common in parrots with chronic respiratory disease but is not diagnostic because other thoracoabdominal pathology (eg, hepatomegaly, abdominal tumors) may also generate this clinical sign. Generalized pulmonary disease is best assayed with these techniques although specific types of focal lesions may also be sampled (Color 13.49).

Biopsies of the spleen are indicated in persistent systemic diseases where an etiologic diagnosis is lacking, in cases of unexplained splenomegaly and in granulomatous inflammation of the spleen.

Samples of the ventricular serosa and muscularis that include nerve tissue can be valuable in the definitive antemortem diagnosis of neuropathic gastric dilatation (NGD). A minimum of two specimens is obtained from the caudoventral surface of the ventriculus. A site near a branching blood vessel is chosen in an attempt to harvest nervous tissue. The thick ventricular muscularis prevents perforation of the viscus. These sites heal well, as only a portion of the serosa and outer muscularis is required. Ventricular biopsies are preferred for the diagnosis of NGD over proventricular biopsies because the serosa of the ventriculus can be harvested much more safely with less risk of perforation due to the thicker muscularis. The ventriculus is believed to be the most important site for NGD involvement due to its role in the motility of the gastrointestinal system. Biopsy of the proventriculus is contraindicated due to its thin wall and the difficulty in preventing perforation with gastric spillage and peritonitis. Some researchers are investigating the possibility of identifying histopathologic lesions of NGD in biopsies of the crop.

Small, precise biopsies of the testicle may be useful in the documentation of reproductive failure due to dysfunction of the testes. Local and systemic infections may cause testicular lesions, although these have been poorly documented.

Contraindications

The specific contraindications for biopsy relate to blood clotting. Biopsy collection should be delayed in any avian patient that shows evidence of abnormalities of the hemostatic system. This usually becomes evident at the time of blood collection. Most birds should show clot formation in one to two minutes. Deficiency of vitamin K is the most common coagulation disorder, for which vitamin K_1h is administered pre-surgically (see Chapter 18). The blood film should be examined for the presence of adequate thrombocyte numbers.

A biopsy cup shape and diameter appropriate to the size of the patient and organ to be biopsied must be chosen. Forceps too large for the purpose may cause excessive organ trauma and hemorrhage.

Biopsy cups generally come in only two shapes: round or elliptical. The round shape does not penetrate as deeply into tissue as the same diameter elliptical cup and this may be indicated for use with certain organs such as the kidney or testes.

Inexperience with the instrumentation and approaches to the organ is a potential cause of biopsy complications.

Instrumentation

Table 13.4 lists selected sources of endoscopic and biopsy equipment.

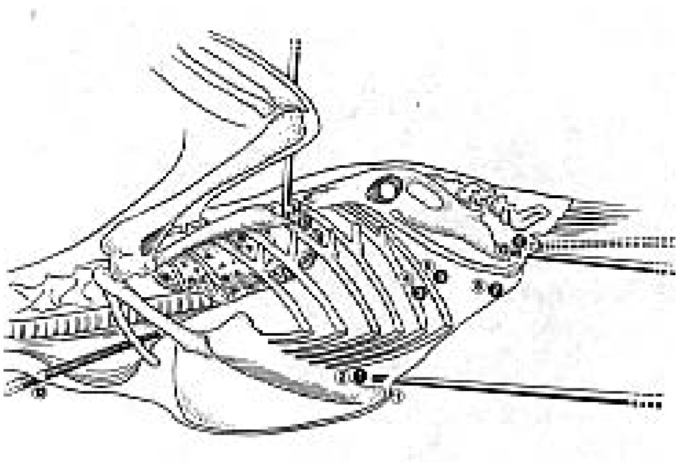
TABLE 13.4 Manufacturers of Endoscopic Equipment

Karl Storz Veterinary Endoscopy-America, Inc. 175 B Cremona Drive Goleta, California 93117 USA
Richard Wolf Medical Instruments Corp. 7046 Lyndon Avenue Rosemont, Illinois 60018 USA
Olympus Corporation 4 Nevada Drive Lake Success, New York 11042-1179 USA
Orlux Engineering and Instrumentation Ltd. 18 Strathearn Avenue, Unit 17 B Brampton, Ontario L6T 4X9 CANADA

A percutaneous technique to biopsy the avian liver using a 19 ga modified Jamshidi or Menghini needle has been described.[18] The larger, right lobe of the liver was approached through the sternal notch with the needle directed posteriorly to avoid puncture of the heart. The technique is relatively rapid to perform, but it is a blind procedure. The liver cannot be inspected nor can focal lesions be sampled. The proventriculus, heart and bile duct are at risk for organ trauma. Optically guided biopsies of the liver are superior.[20] A rigid cup biopsy forceps originally designed for otolaryngology[i] can be guided along the shaft of the endoscope to the visual field. Biopsies of the liver, kidney, spleen, air sacs, lung and testes can be obtained under direct observation. The 8150.00 forceps has a 3.0 mm (9 Fr) diameter cup and takes a relatively large, elliptical sample. This instrument should not be used in birds under 200 g. This forceps in combination with a 2.2 or 2.7 mm telescope has been the most widely utilized and accepted method for collecting optically guided biopsies in the avian patient. The forceps is "walked" into position along the shaft of the endoscope until it can be visualized.

In an effort to improve the usefulness of endoscopically guided biopsies, a new endoscope and sheath set has been developed[a] in cooperation with Karl Storz Endoscopy. An instrument channel permits the use of implements up to 5 Fr (1.7 mm) diameter. Flexible forceps for biopsy and grasping as well as aspiration and infusion cannulas can be placed into the port of the channel and guided easily to the tip of the sheath and into the viewing field of the endoscope. This single puncture system simplifies the manipulation of instrumentation for the endoscopist and helps prevent additional patient trauma.

The system is appropriate for patients weighing approximately 150 g to 2000 g. In larger birds or at certain sites (eg, the ventriculus), a heavier biopsy forceps (eg, 7 Fr) is frequently required. This necessitates a larger sheath. The advantage of a systematic approach to endoscopic equipment employing one manufacturer is that a modular design can be



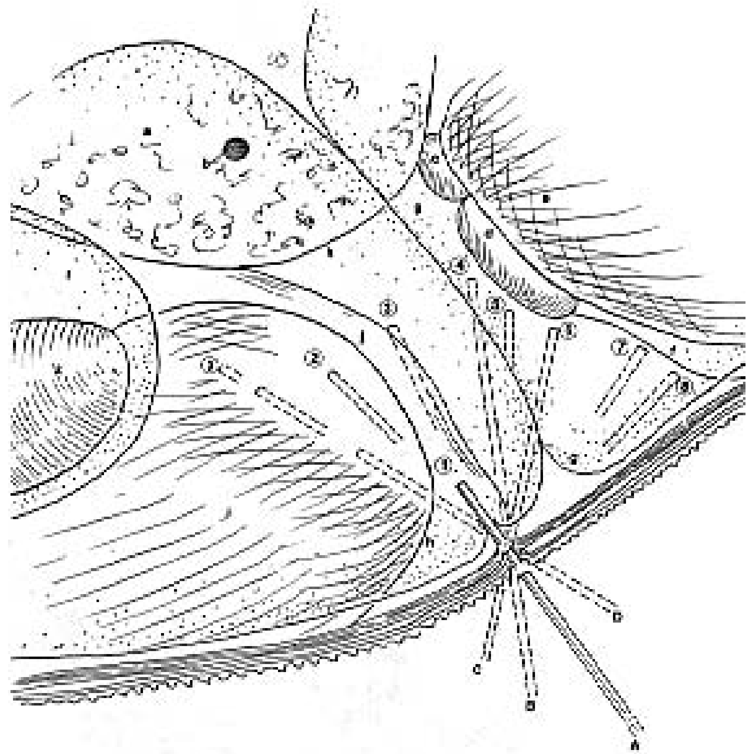


FIG 13.2 *(repeated)* Numbered endoscopic sites described for evaluation of the internal anatomy of birds. Entry sites are shown as either left-sided approaches (open) or right-sided approaches (solid).

FIG 13.4 *(repeated)* The endoscopist can develop an insight into the relative position of organs as viewed from entry site 6. The views are divided into four angles (A,B,C,D) and depths (1 through 9). Structures used for orientation include: a) lung b) ostium of the cranial thoracic air sac c) adrenal gland d) gonad e) kidney f) ureter, oviduct, vas deferens area g) abdominal air sac h) caudal thoracic air sac i) liver j) proventriculus k) heart and l) cranial thoracic air sac.

Endoscopic Examination and Biopsy Techniques

Color 13.22
Gross view from the end of the sternum in a cockatoo placed in dorsal recumbency. The sternum (s) has been elevated to accentuate the division of the cavities visible from endoscope insertion point 11 (Figure 13.2). The confluent wall of the right cranial thoracic air sac and right ventral hepatic peritoneal cavity (1), ventral mesentery (2) and confluent wall of the left cranial thoracic air sac and left ventral hepatic peritoneal cavity (3) are clearly visible. The right liver lobe (rl) and left liver lobes (ll) are also visible. The right ventral hepatic peritoneal cavity is marked by arrows; the left ventral hepatic peritoneal cavity is marked with open arrows.

Color 13.23
(Insertion point 2 see Figure 13.2) View inside the left cranial thoracic air sac of an Amazon parrot. For reference purposes, insertion point 2 would provide a similar view to position D-9 if entering through site 6 as shown in Figure 13.4. Easily identifiable structures include ribs (r), proventriculus (p), medial intercostal muscle (m), heart (h), attachment of pericardial sac (arrow), lung (lu), ostium of cranial thoracic air sac (open arrow) and liver (li).

Color 13.24
(Insertion Point 2 see Figure 13.2) The endoscope is in the cranial thoracic air sac, and the contiguous wall between the cranial and caudal thoracic air sacs is visible caudally (a). In this entry site, the heart (h) will be observed beating cranially. Other structures that can be visualized include ribs (r), lung (lu), liver (l), medial intercostal muscle (m) and the ostium for the cranial thoracic air sac (arrow).

Color 13.25
(Insertion point 10 see Figure 13.2) An endoscope placed in the left abdominal air sac was used to take a picture of a second endoscope guided into the intestinal peritoneal cavity. Note the membrane (arrow) covering the tip of the endoscope with the intestinal (in) tract under the membrane. Other visible structures include the lung (lu), cranial pole of the left kidney (k), transverse abdominal muscle (m), ilium (i) and proventriculus (p).

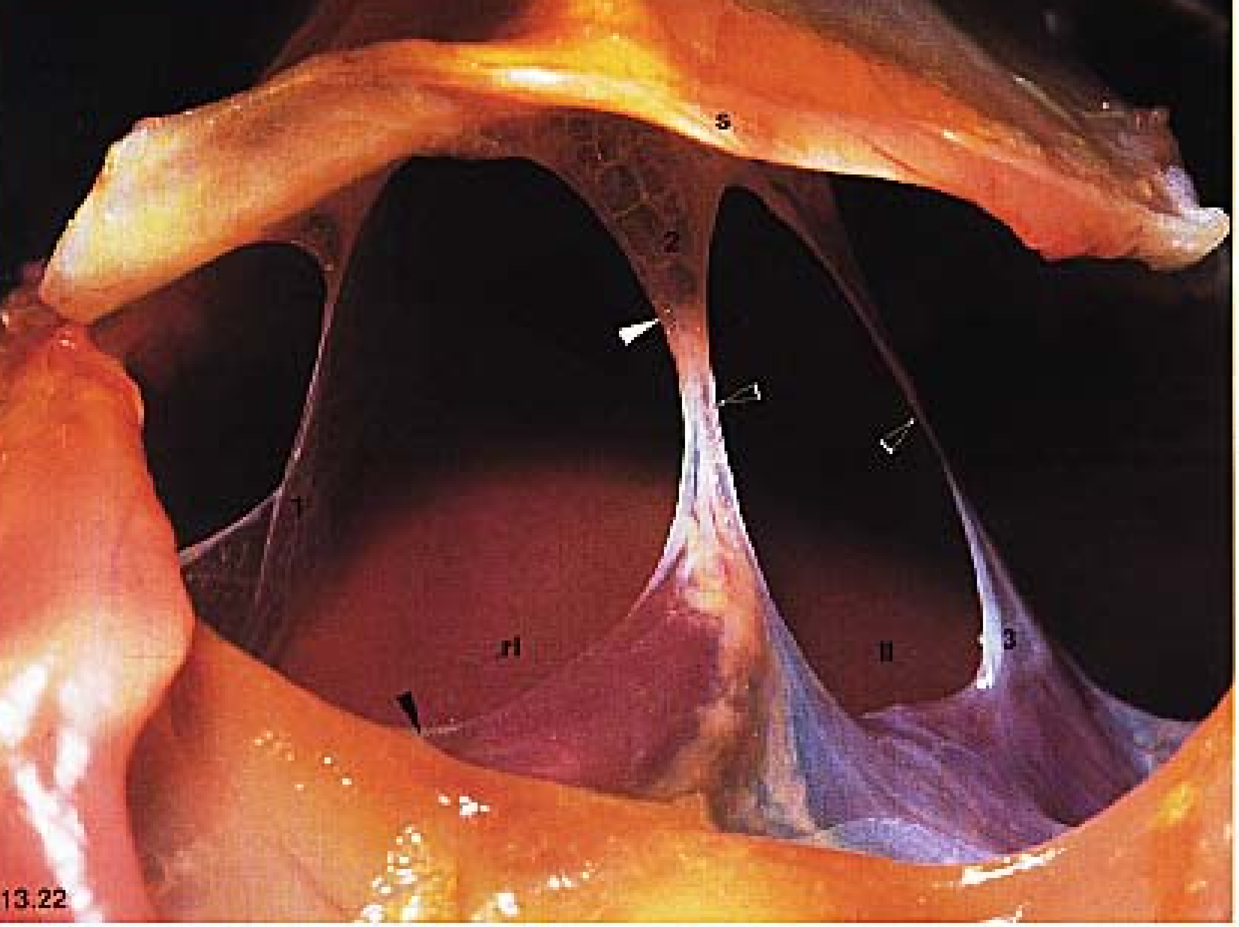
Color 13.26
(Insertion point 10 see Figure 13.2) This Amazon parrot had been endoscoped from insertion point 6 and the iatrogenic tear that was made in the contiguous wall of the caudal thoracic and abdominal air sac is clearly visible (arrow). Equipment used for endoscopy must be sterile to prevent air sac infections or peritonitis. The air sacs were originally clear and now are considered cloudy, and there is an increase in vascularization. When viewed from insertion point 8, a granuloma is evident in the air sac (g). Other structures that are visible include lung (lu), ilium (i), cranial pole of the left kidney (k), loop of intestines (in), proventriculus (p), external iliac vein (open arrow).

Color 13.27
(Insertion point 10 see Figure 13.2) A rent is visible in the contiguous wall of the caudal thoracic and abdominal air sac (arrow) showing the path of the endoscope when inserted at point 6 for gender determination. Other visible structures include lung (lu), external iliac vein (open arrow), cranial division of the left kidney (k1), middle division of the left kidney (k2), (i) ilium, loops of intestines (in) and proventriculus (p). This represents how a site should appear if the original entry was performed under aseptic conditions. Compare this to Color 13.26.

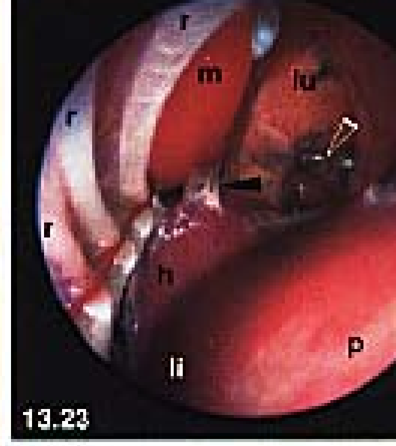
Color 13.28
(Insertion point 11 see Figure 13.2) The normal endoscopic anatomy of the ventral hepatic peritoneal cavity of a pigeon. This view provides clear visualization of the size, shape and texture of the liver. This position can be used to obtain endoscopically guided biopsies of the liver. Note that the right lobe of the liver (rl) extends further caudally than the left lobe of the liver (ll). Other structures that can be visualized include the sternum (s), deep pectoral muscle (m), proventriculus (p) and heart (h).

Color 13.29
A small tear (arrow) has been created in the caudal thoracic air sac to enter the underlying left ventral hepatic peritoneal cavity of a normal pigeon. Liver (li), proventriculus (p), lung (lu), ostium of caudal thoracic air sac (o), contiguous wall of the caudal thoracic and abdominal air sac (a), contiguous wall of the cranial and caudal thoracic air sac (open arrows).

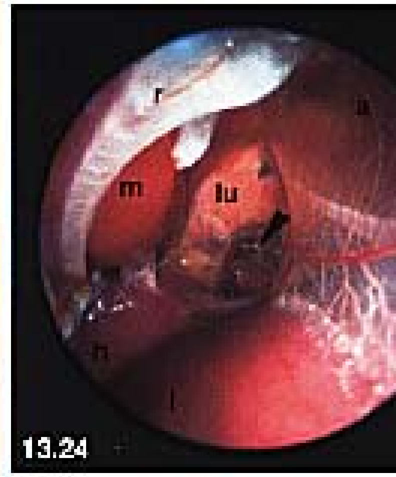
Color 13.30
Left, ventral, hepatic peritoneal cavity distended with air (arrows) following biopsy of the liver. Other visible structures include the lung (lu) and proventriculus (p).



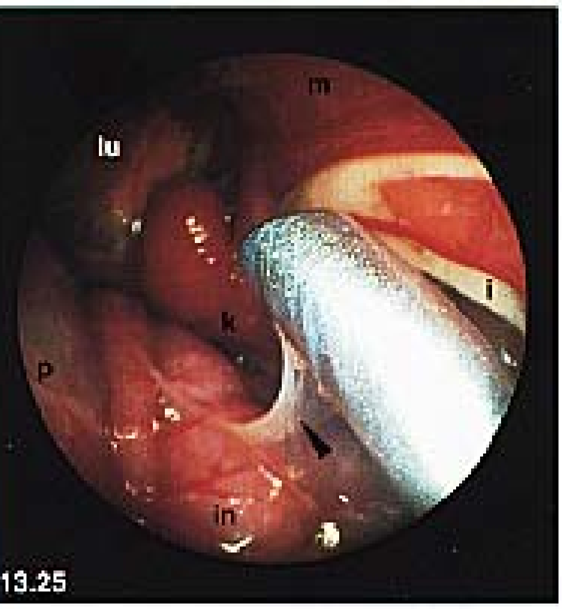
13.22



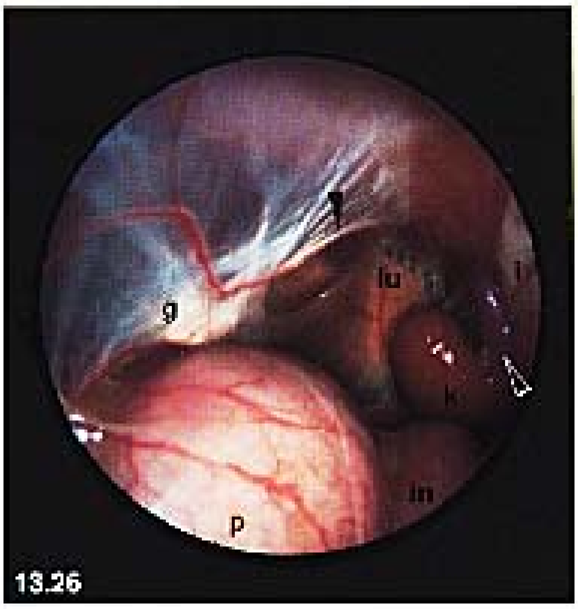
13.23



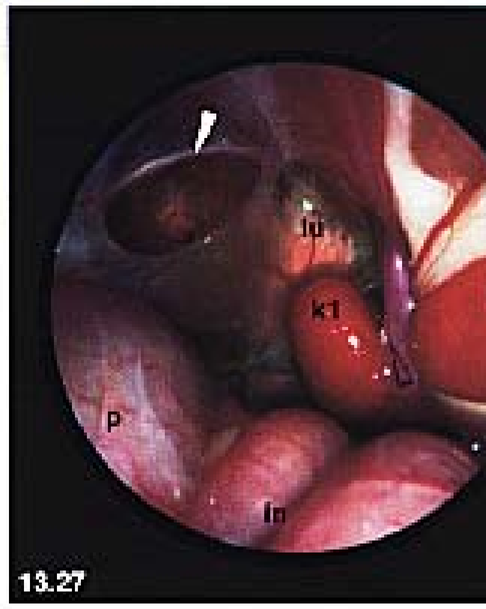
13.24



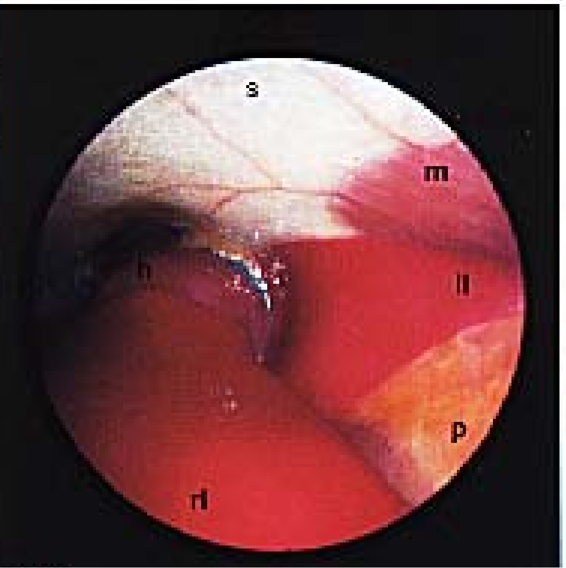
13.25

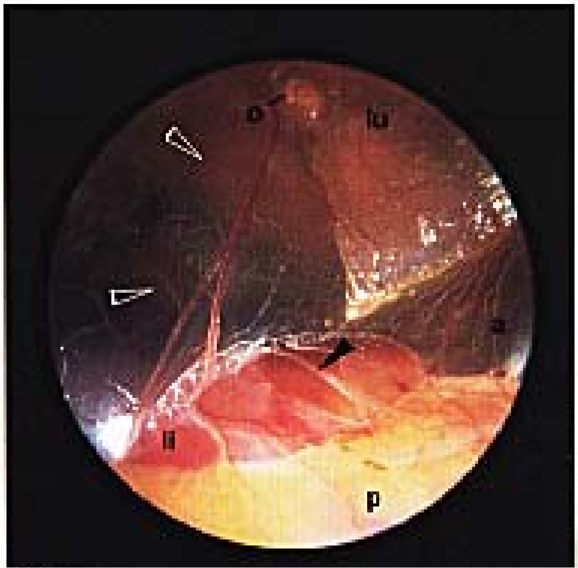


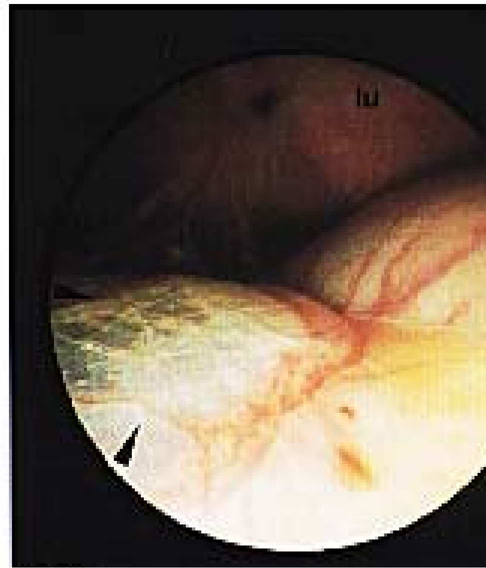
13.26

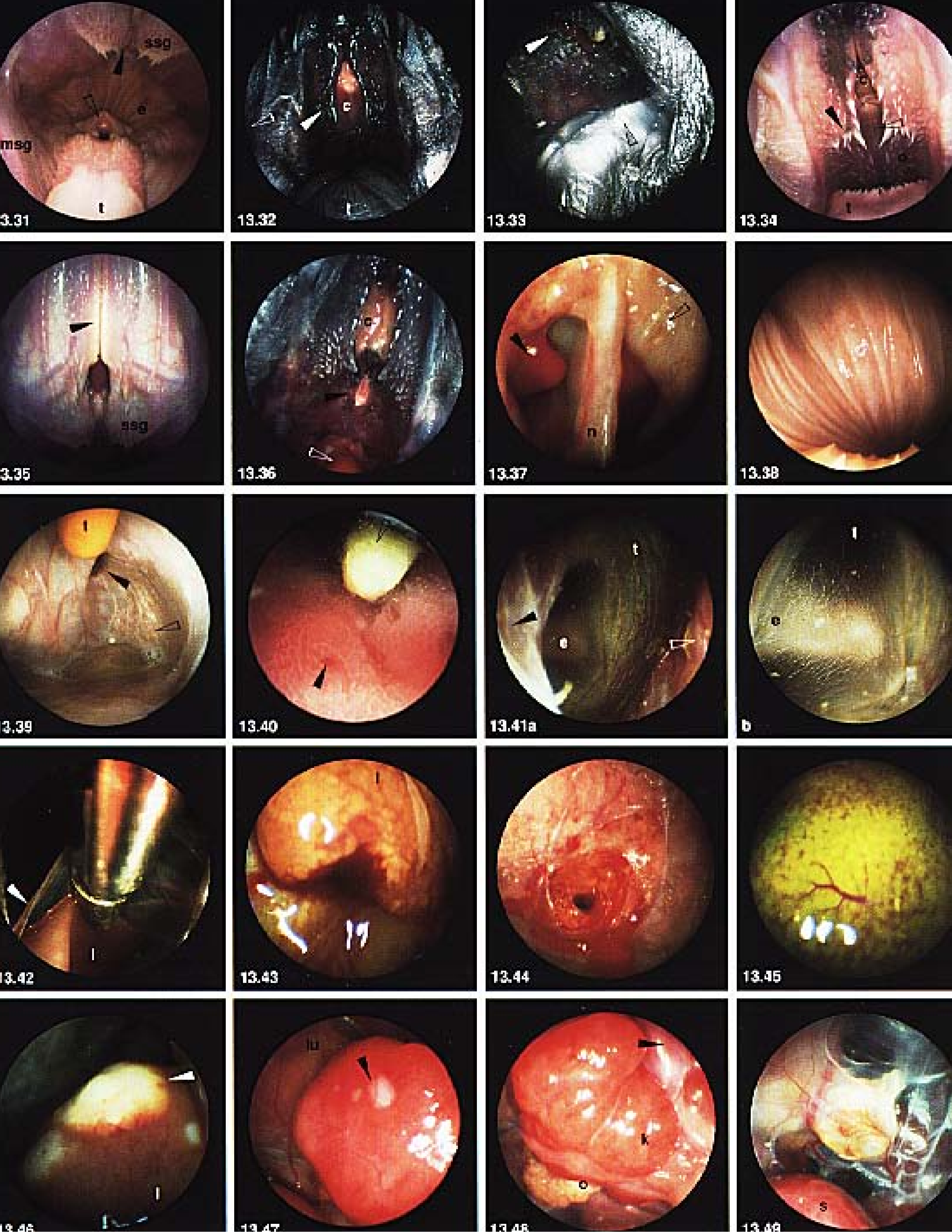


13.27









ssg
e
msg
t
13.31
c
13.32
13.33
c
o
t
13.34
ssg
13.35
c
13.36
n
13.37
13.38
13.39
13.40
t
e
13.41a
e
b
13.42
13.43
13.44
13.45
lu
13.47
k
o
s
13.49

Endoscopic Examination and Biopsy Techniques

Color 13.31
An endoscope has been placed in the oral cavity of a Great Horned Owl showing the infundibular cleft (arrow), sphenopterygoid salivary glands (ssg), longitudinal folds of the esophagus (e), laryngeal mound (open arrow), tongue (t) and mandibular salivary glands (msg).

Color 13.32
An endoscope has been placed in the oral cavity of a normal African Grey Parrot. Note the dark pigmentation and uniform coloration and texture of the oral mucosa. Well formed papillae (arrow) are noted on either side of the choanal slit (c). Also visible are the lateral commissures of the mouth (open arrows) and the tongue (t).

Color 13.33
Lateral view of the oral cavity in a Yellow-crowned Amazon Parrot with hypovitaminosis A. Abscessation (open arrow) at the base of the tongue (t), and blunting and abscessation of the choanal papillae (arrow) are characteristic. Hyperkeratosis of the tongue and oral mucosa are also noted.

Color 13.34
An endoscope has been placed in the oral cavity of a normal Amazon parrot. Note the smooth texture of the tissues in the oral mucosa. All secretions are serous in nature. The choanal (arrow) and lingual papillae are sharp and well defined. Other structures that can be visualized include the tongue (t), oropharynx (o), infundibular cleft (open arrow) and choana (c).

Color 13.35
Endoscopic view of the palate in a Great Horned Owl. The cranial choanal slit (arrow) and sphenoptergoid salivary glands (ssg) are visible. Note that the choanal slit does not contain papillae, but that papillae are present on the caudal edge of the sphenopterygoid salivary glands.

Color 13.36
Caudal view of the choanal area in an African Grey Parrot. The visible structures include the choanal slit (c), infundibular cleft (arrow) and endotracheal tube placed in the trachea (open arrow). Note that the structure of the choanal papillae is different in an African Grey Parrot than in an Amazon parrot (see Color 13.34).

Color 13.37
Endoscopic view of the cranial margin of the choanal slit in a Moluccan Cockatoo. The nasal septum (n), left middle nasal concha (arrow) and nasal mucous membranes (open arrow) are visible.

Color 13.38
Normal esophagus of a Great Horned Owl showing longitudinal folds.

Color 13.39
Normal crop of a cockatoo. A red rubber feeding catheter (t) has been introduced into the crop and is just ventral to the opening from the crop into the thoracic esophagus (arrow). Normal, clear, bubbly mucus is seen covering the crop mucosa. Wrinkling of the crop mucosa (open arrow) is occurring in response to a peristaltic wave. Note the smooth, thin texture and even color of the crop mucosa.

Color 13.40
An endoscope has been passed into the fluid-filled proventriculus of a pigeon. Note the openings of the proventricular glands (arrow) and a pelleted food particle (open arrow).

Color 13.41
a) An endoscope has been inserted into the external ear canal of a Great Horned Owl to show the tympanic membrane (t), extracolumellar cartilage (e), cranial wall of the ear canal (arrow) and caudal wall of the ear canal (open arrow). **b)** Closer view of extracolumellar cartilage.

Color 13.42
Biopsy forceps are being used to take a sample from the caudal edge of the left liver lobe (l). The confluent wall of the caudal thoracic air sac and left ventral hepatic peritoneal cavity membrane are clearly visible (arrow).

Color 13.43
Post-biopsy view of the left liver lobe (l) in a pionus parrot with avian mycobacteriosis.

Color 13.44
Insertion 6. Post-biopsy photograph of the lung as viewed from within the left caudal thoracic air sac.

Color 13.45
Liver of an Amazon parrot showing severe biliverdin accumulation secondary to chlamydiosis.

Color 13.46
Endoscopic view of the liver (l) prior to biopsy of several white-to-yellow proliferative masses (arrow). Histopathology indicated bile duct carcinoma in an Amazon parrot. This bird had a history of cloacal papillomatosis.

Color 13.47
(Position B-4 see Figure 13.4) Endoscopic view of the cranial pole of the left kidney in an Amazon parrot showing several white, proliferative masses (arrow). Biopsy indicated lymphosarcoma. Other visible structures include the lung (lu).

Color 13.48
(Insertion point 6, postion B-4 see Figure 13.4) Endoscopic appearance of chronic glomerulonephritis in an Amazon parrot. The cranial pole of the left kidney (k), common iliac vein (arrow) and ovary (o) are clearly visible.

Color 13.49
Granuloma in the abdominal air sac of a pionus parrot. The substantial vascularity of the adjacent air sacs suggests a chronic reaction. The fact that the air sac tissue at the periphery of the mass is normal suggests that the infection has been contained. The spleen (s) is enlarged and pale.

used. Thus the 2.7 mm endoscope in the standard Storz avian set can be inserted into other sheaths such as the modified 26156 H, which permits the introduction and use of the larger 7 Fr biopsy forceps.

Preparation of Small Biopsies

The biopsies obtained with the types of forceps previously mentioned are small and must be handled with care so that they are not lost or damaged. Various techniques have been recommended in the past to enable the histotechnologist to locate and properly imbed small specimens for processing. Wrapping tiny pieces of tissue in filter paper or a very fine cloth before immersion in the fixative is one method. Or the specimens can be placed into a small stoppered blood collection container without anticoagulant.[k] This system is simple and effective, allowing the technician to clearly visualize the sample(s). No more than two to three specimens should be placed in each clearly labelled container. Small tissue samples require far less time to fix than larger samples (likely less than two hours in formalin). Specifically buffered, ten per cent formalin designed for tissue fixation must be used. Failure to do so will lead to precipitates and artifacts. If biopsies cannot be processed immediately, the specimens can be stored in a solution of 97% methyl alcohol after fixation in order to ensure sample quality. The laboratory should be contacted for specific recommendations.

Consulting Pathologists

The value of the clinical biopsy is directly related to the quality of the sample, the history provided and the experience of the pathologist. Reading small surgical biopsies from exotic avian species is a relatively specialized area of pathology. Best results are likely to be obtained by working with a consultant pathologist who has a real interest and expertise in this field. Timely reporting of results is essential to enable the clinician to make optimal use of the biopsy information.

Products Mentioned in the Text

a. Avian Endoscopy Diagnostic Set, 2.7 mm, Karl Storz Veterinary Endoscopy, Goleta, CA, 64108 BS, 2.7 mm, 30 Telescope, 67065 C Sheath, 67161 Z Biopsy Forceps.
b. Focuscope, MDS Inc, Clearwater, FL
c. Gluterex, 3M Medical Products, St. Paul, MN.
d. Tegaderm 1626 and 9505, 3M Medical-Surgical Division, St. Paul, MN, OpSite 4963C, Smith and Nephew Inc., Lachine, QC
e. AErrane, Anaquest, Madison, WI; Isoflo, Solvay Animal Health, Mendota Heights, MN
f. Vetwrap, 3M Corp, St. Paul, MN
g. Sovereign, Sherwood Medical, St. Louis, MO
h. AquaMephyton, MSD, Rahway, NJ
i. Forceps 8150.00, Richard Wolf Medical, Rosemont, IL
j. Storz # 27071 T, Karl Storz Veterinary Endoscopy, Goleta, CA
k. Vacutainer red top three ml. tube #6381, Becton Dickinson, Rutherford, NJ

References and Suggested Reading

1. **Bailey RE:** Surgery for sexing and observing gonad condition in birds. Auk 70:497-499, 1953.
2. **Bush M:** Laparoscopy in birds and reptiles. *In* Harrison RM, Wildt DE (eds): Animal Laparoscopy. Baltimore, Williams and Wilkins, l980, pp 183-197.
3. **Degernes LA, et al:** Lead poisoning in trumpeter swans. Proc Assoc Avian Vet, , l989, pp l44-l55.
4. **Evans HE:** Anatomy of the budgerigar. *In* Petrak ML (ed): Diseases of Cage and Aviary Birds 2nd ed. Philadelphia, Lea and Febiger, 1982, pp 111-187.
5. **Frankenhuis MT, Kappert HJ:** Infertility due to surgery on body cavity in female birds: Cause and prevention. XXII Inter Symp Erkr Zoo-Arnhem, Akademie-Verlag, Berlin, 1980, pp 237-239.
6. **Goodchild WM:** Differentiation of the body cavities and air sacs of *Gallus domesticus* postmortem and their location *in vivo*. Br Poult Sci ll:209-215, l970.
7. **Harrison GJ:** Endoscopic examination of avian gonadal tissue. Vet Med Small Anim Clin 73:479-484, l978.
8. **Harrison GJ:** Endoscopy. *In* Harrison GJ, Harrison LR (eds): Clinical Avian Medicine and Surgery. Philadelphia, WB Saunders Co, l986, pp 224-244.
9. **Hopkins HH:** Optical principles of the endoscope. *In* Berci G (ed): Endoscopy. New York, Appleton Century Crofts, 1976, pp 3-27.
10. **Hulka JF:** Textbook of Laparoscopy. Orlando, Grune and Stratton, 1985, p 11.
11. **Hunter DB, Taylor, M:** Lung biopsy as a diagnostic technique in avian medicine. Proc Assoc Avian Vet, 1992, pp 207-211.
12. **Ingram KA:** Laparotomy technique for sex determination of psittacine birds. J Am Vet Med Assoc l73(9):1244-1246, 1978.
13. **Ingram KA:** Otoscopic technique for sexing birds. *In* Kirk RW (ed): Current Veterinary Therapy VII. Philadelphia, WB Saunders Co, 1980, pp 656-658.
14. **Johnson AL:** Reproduction in the female and male. *In* Sturkie PD (ed): Avian Physiology 4th ed. New York, Springer Verlag, 1986, p 404.
15. **Jones DM, et al:** Sex determination of monomorphic birds by fibreoptic endoscopy. Vet Rec 115:596-598, 1984.
16. **King AS, McLelland J:** Female reproductive system. *In* Birds: Their Structure and Function. London, Balliére Tindall, 1984, pp l45-l65.
17. **King AS, McLelland J:** Respiratory system. *In* Birds: Their Structure and Function. London, Balliére Tindall, 1984, pp 110-144.
18. **Kollias GV:** Liver biopsy techniques in avian clinical practice. Vet Clin North Am l4(2):287-298, 1984.
19. **Kollias GV:** Avian endoscopy. *In* Jacobson ER, Kollias GV (eds): Contemporary Issues in Small Animal Medicine, Exotic Animals. New York, Churchill Livingstone, 1988, pp 75-104.
20. **Kollias GV, Harrison GJ:** Biopsy techniques. *In* Harrison GJ, Harrison LR (eds): Clinical Avian Medicine and Surgery. Philadelphia, WB Saunders Co, 1986, pp 245-249.
21. **Lumeij JT:** Endoscopy. A Contribution to Clinical Investigative Methods for Birds with Special Reference to the Racing Pigeon (*Columbia livia domestica*). Utrecht, PhD Thesis, 1987, pp l51-166.
22. **McDonald SE:** Surgical sexing of birds by laparoscopy. Calif Vet 5:l6-22, l982.
23. **McDonald SE:** Endoscopic examination. *In* Burr EW (ed): Companion Bird Medicine. Ames, Iowa State University Press, 1987, pp 166-174.
24. **McLelland J, King AS:** The gross anatomy of the peritoneal and coelomic cavities of *Gallus domesticus*. Anat Anaz Bd 127:480-490, l970.
25. **Redig PT:** Aspergillosis. *In* Kirk RW (ed): Current Veterinary Therapy VIII. Philadelphia, WB Saunders Co, 1983, pp 611-613.
26. **Risser AC:** A technique for performing laparotomy on small birds. Condor 73:376-379, 1971.
27. **Satterfield W:** Diagnostic laparoscopy in birds. *In* Kirk RW (ed): Current Veterinary Therapy VII. Philadelphia, WB Saunders Co, 1980, pp 659-661.
28. **Satterfield W:** Early diagnosis of avian tuberculosis by laparoscopy and liver biopsy. *In* Cooper JE, Greenwood AG (eds): Recent Advances in the Study of Raptor Diseases. Keighley, Chiron Publications, 1981, pp l05-l06.
29. **Taylor M:** A morphologic approach to the endoscopic determination of sex in juvenile macaws. J Assoc Avian Vet 3(4):l99-201, 1989.
30. **Taylor M:** Endoscopy. Proc Assoc Avian Vet, Phoenix, 1990, pp 319-323.
31. **Taylor M:** Endoscopy. Laboratory Manual. Assoc Avian Vet, 1992, pp 1-10.