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ROUTINE PROCEDURES

ADMINISTRATION TECHNIQUES FOR MEDICATIONS AND FLUIDS

ORAL ADMINISTRATION: TABLETS AND CAPSULES—CANINE

Patient Preparation
None required.

Technique
The simplest method of administering tablets or capsules to dogs is to hide the medication as bait in food. Offer small portions of unbaited cheese, meat, or some favorite food to the dog initially. Then offer one portion that includes the medication. Pill Pockets Canine Treats* is a commercially available alternative.

*Pill Pockets Treats for Dogs and Pill Pockets Treats for Cats; Greenies (www.greenies.com).
For anorexic dogs or when pills must be given without food, give medications quickly and decisively so that the process of administering the medication is accomplished before the dog realizes what has happened. With cooperative dogs, insert the thumb of one hand through the interdental space, and gently touch the hard palate. This will induce the cooperative dog to open its mouth (Figure 4-1). Using the opposite hand (the one holding the medication), gently press down on the mandible to open the mouth further (Figure 4-2).

**Figure 4-1:** Use of the thumb only to open a cooperative dog’s mouth.

**Figure 4-2:** Use of the opposite hand to place a tablet or capsule on the caudal aspect of the tongue.
Quickly place the tablet or capsule onto the caudal aspect of the tongue. Quickly withdraw the hand and close the dog’s mouth. When the dog licks its nose, the medication likely has been swallowed.

Dogs that offer more resistance can be induced to open their mouths by compressing their upper lips against their teeth. As they open the mouth, roll their lips medially so that if they attempt to close the mouth, they will pinch their own lips. Alternatively, dripping water onto the nostrils or blowing into the patient’s nose sometimes encourages the patient to accept and swallow oral medications (tablets or capsules). Pilling syringes are also available and in some dogs seem to work well.

**Special Considerations**
Critical to the oral administration of medication is the ability of the owner to effectively administer the medication at home. Animals that aggressively resist oral medication should be treated by alternative methods—for example, parenteral administration of medication. It is inappropriate, and unsafe, to delegate treatment responsibilities to the owner of a dog (or cat) that might injure the individual who is attempting to treat the patient.

**Oral Administration: Tablets and Capsules—Feline**

*Patient Preparation*
None required.

*Technique*

*Caution:* Only experienced individuals should attempt this technique of administering tablets or capsules to cats. Even cooperative cats that become intolerant will bite. Therefore, this is not a technique recommended for most owners to try at home, even if specific instructions have been given.

Two methods of pill administration are used in cats. In both methods the cat’s head is elevated slightly with the nose pointed upward. Success in administering pills and tablets to a cat entails a delicate balance between what works well and what works safely. In cooperative cats, it may be possible to use one hand to hold and position the head (Figure 4-3) while using the opposite hand (the one holding the medication) to open the mouth gently by depressing the proximal aspect of the mandible (Figure 4-4). Press the skin adjacent to the maxillary teeth gently between the teeth as the mouth opens, thereby discouraging the
cat from closing its mouth. With the mouth open, drop (do not push) the medication (try generously lubricating the tablet or capsule with butter) into the oral cavity. The cat can be tapped under the jaw or on the tip of the nose to facilitate swallowing if you really think this works. If the cat licks, administration was probably successful.

Alternatively, some cats will tolerate a specially designed “pilling syringe” in an attempt to administer a tablet or capsule. The pilling syringe works well as long as it is inserted cautiously and atraumatically into the cat’s mouth. However, if resistance ensues, the rigid pilling syringe may injure the hard palate during the ensuing struggle. Subsequent attempts to use the syringe may be met with increasing resistance and increasing risk of injury. Success with a pilling syringe depends largely on the cat. Pill Pockets Treats are also available for use in cats and are manufactured in chicken and fish flavors. In addition, as is the case in dogs, some cats will respond to the application of water drops on the nostrils or blowing into the nostrils to encourage swallowing.

Special Considerations
When dispensing oral medications for home administration to cats, do not expect clients to force a tablet or capsule into a cat’s mouth. Although some clients are remarkably capable and confident with their ability to administer oral medications to cats, the risk of injury to the client can be significant. Whenever feasible, liquid medications or pulverized tablets should be mixed with the diet or an oral treat readily accepted and consumed (see the following discussion).

**Oral Administration: Liquids**

**Without a Stomach Tube**

*Patient Preparation*
None required. Technique is appropriate for owners to perform at home.

*Technique*
Small amounts of liquid medicine can be given successfully to dogs and cats by pulling the commissure of the lip out to form a pocket (Figure 4-5). Deposit the liquid medication into the “cheek pouch,” where it subsequently flows between the teeth as the head is held slightly upward. Patience and gentleness, along with a reasonably flavored medication, contribute to the success.
Spoons are ineffective, as fluids are easily spilled. A disposable syringe can be used to measure and administer liquids orally. Depending on the liquid administered, disposable syringes can be reused several times, assuming they are rinsed after each administration. In addition, disposable syringes can be dispensed legally to clients for home administration of liquid medication. Mixing of medications in the same syringe is not recommended. However, dispensing of a separate, clearly marked syringe for each type of liquid medication prescribed for home administration is recommended.

**Special Considerations**
Compounding pharmacies are also available and can mix many medications into palatable flavors to help facilitate the oral administration of medications.

Dogs with swallowing disorders should not be treated at home with liquid medications because this could cause complications associated with aspiration.

**With an Administration Tube**

*Patient Preparation*
None required.

*Note:* This procedure is reserved for in-hospital use only. The technique should be performed only by individuals trained to perform this procedure.

*Technique*
Administration of medications, contrast material, and rehydrating fluids can be accomplished with the use of a well lubricated feeding tube passed through the nostrils into the stomach or distal esophagus. When a feeding tube is placed for long-term use (multiple days) and repeated use (described under Gastrointestinal Procedures later), it is generally recommended to avoid passing the tip of the tube beyond the distal esophagus. The reason for recommending nasoesophageal intubation over nasogastric intubation is based on the fact that reflex peristalsis of the esophagus against a tube passing through the cardia can result in significant mucosal ulceration within 72 hours. This is not a factor in patients receiving a single dose of medication or contrast material.
The narrow lumen of tubes passed through the nostril of small dogs and cats limits the viscosity of solutions that can be administered through a tube directly into the gastrointestinal tract. Nasoesophageal intubation can be done with a variety of tube types and sizes (Table 4-1). Newer polyurethane tubes, when coated with a lidocaine lubricating jelly, are nonirritating and may be left in place with the tip at the level of the distal esophagus. When placing the nasogastric tube, instill 4 to 5 drops of 0.5% proparacaine in the nostril of the cat or small dog; 0.5 to 1.0 mL of 2% lidocaine instilled into the nostril of a larger-breed dog may be required to achieve the level of topical anesthesia needed to pass a tube through the nostril. With the head elevated, direct the tube dorsomedially toward the alar fold (Figure 4-6). Pushing dorsally on the nasal philtrum and pushing the nostril from lateral to medially will help facilitate passage of the tube into the ventromedial nasal meatus.

Caution: The tip of the feeding tube can be inadvertently introduced through the glottis and into the trachea. Topical anesthetic instilled into the nose can anesthetize the arytenoid cartilages, thereby blocking a cough or gag reflex.

After inserting the tip 1 to 2 cm into the nostril, continue to advance the tube until it reaches the desired length. If the turbinates obstruct the passage of the tube, withdraw the tube by a few centimeters. Then readvance the tube, taking care to direct the tube ventrally through the nasal cavity. Occasionally it will be necessary to withdraw the tube completely

*Multiple types of pediatric polyurethane nasogastric feeding tubes are available in sizes ranging from 8F to 12F that easily accommodate administration of liquids medications and fluids to kittens, cats, and small dogs.

| Size | Millimeters | Inches |
|------|-------------|--------|
| 3    | 1           | 0.039  |
| 4    | 1.35        | 0.053  |
| 5    | 1.67        | 0.066  |
| 6    | 2           | 0.079  |
| 7    | 2.3         | 0.092  |
| 8    | 2.7         | 0.105  |
| 9    | 3           | 0.118  |
| 10   | 3.3         | 0.131  |
| 11   | 3.7         | 0.144  |
| 12   | 4           | 0.158  |
| 13   | 4.3         | 0.170  |
| 14   | 4.7         | 0.184  |
| 15   | 5           | 0.197  |
| 16   | 5.3         | 0.210  |
| 17   | 5.7         | 0.223  |
| 18   | 6           | 0.236  |
| 19   | 6.3         | 0.249  |
| 20   | 6.7         | 0.263  |
| 22   | 7.3         | 0.288  |
| 24   | 8           | 0.315  |
| 26   | 8.7         | 0.341  |
| 28   | 9.3         | 0.367  |
| 30   | 10          | 0.393  |
| 32   | 10.7        | 0.419  |
| 34   | 11.3        | 0.445  |
from the nostril and repeat the procedure. In particularly small patients or patients with obstructive lesions (e.g., tumor) in the nasal cavity, it may not be possible to pass a tube. Do not force the tube against significant resistance through the nostril.

Gavage, or gastric lavage and feeding, in puppies and kittens can be accomplished by passing a soft rubber catheter or feeding tube into the mouth, tilting the puppy’s or kitten’s head, and watching it swallow the tube. Most puppies or kittens will struggle and vocalize. They usually will not vocalize if the tube has been placed into the trachea. A 12F catheter is of an adequate diameter to pass freely, but it is too large for dogs and cats less than 2 to 3 weeks of age. Mark the tube with tape or a pen at a point equal to the distance from the mouth to the last rib. Merely push the tube into the pharynx and down the esophagus to the caudal thoracic level (into the stomach). Verify the placement of the tube using the same dry syringe aspiration technique to ensure that the tube is positioned in the esophagus or stomach rather than the trachea. Attach a syringe to the flared end, and slowly inject medication or food.

Depending on the feeding tube type, the end of the tube may or may not accommodate a syringe. For example, soft, rubber urinary catheters are excellent tubes for single administration use. However, the flared end may not accommodate a syringe. To affix a syringe to the outside end of a tapered feeding tube or catheter, insert a plastic adapter (Figure 4-7) into the open end of the tube.

**Figure 4-6:** Initial dorsomedial placement of a nasoesophageal tube before complete insertion.

**Figure 4-7:** Use of a plastic adaptor (“Christmas tree”) to affix a syringe to a nasoesophageal feeding tube.
**ROUTINE PROCEDURES**

**Special Considerations**
Esophageal (versus intratracheal) placement of the feeding tube can be verified with a dry, empty syringe. Attach the empty syringe to the end of the feeding tube. Rather than injecting air or water in an attempt to auscultate borborygmus over the abdomen, attempt simply to aspirate air from the feeding tube. **If there is no resistance during aspiration and air fills the syringe, the tube likely has been placed in the trachea.** Completely remove the tube and repeat the procedure. However, if repeated attempts to aspirate are met with immediate resistance and no air enters the syringe, the tube tip is positioned properly within the esophagus. If there is any question regarding placement, a lateral survey radiograph is indicated.

Definitive confirmation of proper tube placement can be made by diluting 1 to 2 ml of an iodinated contrast agent with sterile saline, instilling the liquid into the tube, then taking a lateral thoracoabdominal radiograph to confirm entry of the contrast material into the stomach.

**TOPICAL ADMINISTRATION**

**Ocular**

*Patient Preparation*

None required.

*Technique*

The usual methods of applying medication directly to the eyes include liquid (drops) and ointments. The route and frequency of medication depend on the disease being treated. Liquids and ointments are appropriate for owner administration.

Liquid medications (usually 1 or 2 drops) can be applied directly to the cornea. It is important to instruct the owner on the proper technique and to stress that because liquids only fall downward, the patient’s nose must be directed upward before one attempts to administer liquid medications onto the eye. It is still quite difficult to encourage a drop of liquid, as it is squeezed from its container, to fall horizontally, despite frequent attempts to do so. Ointment, as a ⅛- or ¼-inch strip, is typically administered directly onto the sclera (dorsally) or into the lower conjunctival cul-de-sac such that as the lids close, a film of ointment is spread across the cornea.

*Special Considerations*

The tip of the applicator tube for liquids and ointments should never be allowed to make contact with the eye or conjunctiva. Doing so is likely to result in contamination of the medication, especially with liquid medications.

**Otic**

*Patient Preparation*

None required.

*Technique*

Liquid solutions are more effective vehicles for administering medication into the external ear canal. Physically removing debris may be necessary in some patients that require topical otic medications. Occasionally, oral supplemental medication may also be required. When applying the medication, a few drops of liquid are generally sufficient. The ear should be massaged gently after instillation to facilitate the spread the medication within the external ear canal.

*Special Considerations*

Medicated powders generally are contraindicated in the external ear canal. Also, the application tip of liquid medications must not come in direct contact with the skin. Doing so is likely to result in contamination of the entire dispensing bottle.
Nasal

Patient Preparation

None required.

Technique

Intranasal administration of liquids in dogs and cats is usually limited to a single dose of a vaccine specifically labeled for intranasal administration. There is little indication for routine instillation of liquids into the nostrils of dogs and cats. Rarely, administration of isotonic solutions directly into the nostrils is indicated. In contrast to single-dose vaccines, lavage solutions applied intranasally are usually multiple-dose containers. Therefore the tip of the administration device should not be allowed to directly contact the patient’s skin or nose. Doing so may result in contamination of the entire bottle. Oily drops are not advised because they may damage the nasal mucosa or may be inhaled.

The technique for intranasal administration of vaccine is straightforward and usually works quite well—the first time. Some animals, dogs more than cats, will aggressively resist intranasal administration of vaccine. Attempts to overcome this resistance include covering the eyes with a towel or otherwise distracting the patient with noise or other visual cues.

Special Considerations

Concerns expressed over the loss of vaccine immediately after intranasal administration are generally unfounded. Manufacturers of intranasal vaccines typically include a greater antigen (virus or bacteria) titer per dose than is necessary to induce a protective immune response. If the patient resists aggressively and the vaccine is indicated, parenteral preparations are available for all intranasal vaccines and should be considered.

Dermatologic

Patient Preparation

Several objectives should be considered when treating dermatologic disorders with topical medication: (1) eradication of causative agents; (2) alleviation of symptoms, such as reduction of inflammation; (3) cleansing and debridement; (4) protection; (5) restoration of hydration; and (6) reduction of scaling and callus. Many different forms of skin medications are available, but the vehicle in which they are applied is a critical factor (Box 4-1).

**BOX 4-1 VEHICLES USED IN THE ADMINISTRATION OF TOPICAL SKIN MEDICATIONS**

- **Lotions** are suspensions of powder in water or alcohol. They are used for acute, eczematous lesions. Because they are less easily absorbed than creams and ointments, lotions need to be applied two to six times a day.
- **Pastes** are mixtures of 20% to 50% powder in ointment. In general, they are thick, heavy, and difficult to use.
- **Creams** are oil droplets dispersed in a continuous phase of water. Creams permit excellent percutaneous absorption of ingredients.
- **Ointments** are water droplets dispersed in a continuous phase of oil. They are very good for dry, scaly eruptions.
- **Propylene glycol** is a stable vehicle and spreads well. It allows good percutaneous absorption of added agents.
- **Adherent dressings** are bases that dry quickly and stick to the lesion.
- **Shampoos** are usually detergents designed to cleanse the skin. If shampoos are left in contact with the skin for a time, added medications may have specific antibacterial, antifungal, or antiparasitic effects.
Technique
In all cases, apply topical medications to a clean skin surface in a very thin film, because only the medication in contact with the skin is effective. In most cases, clipping hair from an affected area enhances the effect of medication. When dispensing medications to owners for home administration, the owner should be instructed to wear disposable examination gloves if using fingers and hands to apply the medication.

Special Considerations
With the widespread availability of compounding pharmacies, prescribing compounded medications for topical and oral administration recently has become a popular dispensing technique for dogs and cats requiring long-term, daily medication. Caution is warranted. Some compounding pharmacies that serve the veterinary profession are using inappropriate or ineffective vehicles in which the drug has been compounded, or the drug itself, purchased in bulk, is of a lower grade and possibly an ineffective product once compounded. Studies on the quality and efficacy of compounded drugs for use in veterinary patients are limited. However, of those studies that have been performed, serious questions are being raised over the bioavailability of the drug administered.

Administration by Injection (Parenteral Administration)

Patient Preparation
It would be admirable to prepare the skin surgically before making needle punctures to administer medications. Because such preparation is not practical, carefully part the hair and apply a high-quality skin antiseptic such as isopropyl alcohol. Place the needle directly on the prepared area, and thrust the needle through the skin. Although the use of antiseptics on the vial and skin is not highly effective, the procedure removes gross contamination and projects an image of professionalism. Before aspirating medications from multiple-dose vials, carefully wipe the rubber diaphragm stopper with the same antiseptic used on the skin. Observe this basic rule with all medication vials, even with modified live virus vaccines.

Subcutaneous Injection

Technique
Dogs and cats have abundant loose alveolar tissue and easily can accommodate large volumes of material in this subcutaneous space. The dorsal neck is seldom used for subcutaneous injections because the skin is somewhat more sensitive, causing some patients to move abruptly during administration. A wide surface area of skin and subcutaneous tissue over the dorsum from the shoulders to the lumbar region makes an ideal site for subcutaneous injections.

Administration of drugs, vaccines, and fluids by the subcutaneous route represents the most commonly used route of parenteral administration in dogs and cats. For small volumes (<2 mL total), such as vaccines, a 22- to 25-gauge needle generally is used. The site most often used is the wide area of skin over the shoulders. The large subcutaneous space and the relative lack of sensitivity of skin at this location make it an ideal injection site. Cleaning of the skin with alcohol or other disinfectant generally is performed before injection. Several injection techniques are used. A common technique entails grasping a fold of skin with two fingers and the thumb of one hand. Gently lift the skin upward. Using the opposite hand, place the needle, with syringe attached, through the skin at a point below the opposite thumb. Aspiration before injection is not typically necessary when using this route of administration. After administration and on removal of the needle from the skin, gently pinch the injection site and hold it for a few seconds to prevent backflow of medication or vaccine onto the skin.

When larger volumes are to be administered—fluids in dehydrated dogs and cats—the skin directly over the shoulders is the injection site most commonly selected. Generally, only isotonic fluids are administered by the subcutaneous route. Depending on the patient's
size, needles ranging from 16 to 22 gauge can be used. Because of the larger volumes of fluid involved, warming of the fluids before administration is recommended. Doing so can enhance significantly the patient's tolerance for the displacement of skin during the period of administration and, in small patients, prevent hypothermia.

Depending on the rate of administration and breed of dog, relatively large volumes of fluid generally can be given in one location. Cats typically tolerate 10 to 20 mL/kg body mass in a single location. Large dogs can tolerate volumes greater than 200 mL of fluid in a single location. When administering large volumes, it is usually not necessary to use multiple injection sites for purposes of distributing the total fluid volume. Doing so actually may increase the risk of introducing cutaneous bacteria under the skin. Because the administration time required to deliver larger volumes is longer, and the injection needle will be placed in the skin for extended periods, it is appropriate to cleanse and rinse the skin carefully before actually inserting the needle. Isotonic, warmed fluids may be administered by large syringe or through an administration tube attached to a bag. Monitor skin tension and the patient's comfort tolerance throughout the procedure.

Although fluid absorption begins almost immediately on subcutaneous administration of fluids, significant pressure caused by the bolus of fluid delivered can develop within the fluid pocket. On removal of the needle, firmly grasp the injection site with the thumb and forefinger for several seconds. The procedure is not complete until one has verified that back-leakage of fluid from the subcutaneous space onto the skin is not occurring. Depending on the patient's hydration status and physical condition, fluid absorption may take from 6 to 8 hours.

Special Considerations
The rate of absorption of fluid administered by the subcutaneous route largely depends on the patient's hydration state and vascular and cardiac integrity. For that reason, the subcutaneous route is not recommended to manage patients in hypovolemic shock. Exceptions to this do exist—for example, when in a life-or-death situation access to a vein is simply not possible. Subcutaneous or intraosseous (see the following discussion) fluid administration may be the only option available.

Implanted Subcutaneous Fluid Ports

Technique
Recently, implantable subcutaneous ports* have been introduced for use in patients requiring regular administration of subcutaneous fluids at home. A 9-inch silicon tube is pre-placed under the skin and is sutured in place by a veterinarian. Objectively, this offers easy access to the subcutaneous space without the need for needle penetration. Owners simply attach a syringe or extension tube tip to the port and administer the appropriate volume of fluids at an appropriate rate and frequency.

Special Considerations
Because of the usual requirement for long-term placement of an implantable fluid administration tube, there is some risk of infection under the skin and around the incision site. Some cats do not tolerate the device.

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*GIF-Tube Single Implant Kit for Subcutaneous Fluid Administration, various models available. Phoenix, Arizona, [www.practivet.com](http://www.practivet.com) (owner instruction guide is also available).
INTRAMUSCULAR INJECTION

Patient Preparation
None required.

Technique
Because the tightly packed muscular tissue cannot expand and accommodate large volumes of injectables without trauma, medications given by the intramuscular route should be small in volume. These medications are often depot materials that are poorly soluble, and some may be mildly irritating. Unless the animal is extremely thin, give injections into the lumbodorsal muscles on either side of the dorsal processes of the vertebral column.

After proper preparation of the skin, insert the needle through the skin at a slight angle (if the animal is thin) or perpendicularly (if the animal is obese). When injecting any medication by a route other than the intravenous one, it is imperative to retract the plunger of the syringe before injecting to be certain that a vein was not entered by mistake. This is especially crucial with oil suspension, microcrystalline suspension, or potent-dose medications.

Special Considerations
Never give intramuscular injections in the neck because of the fibrous sheaths there and the complications that may occur. Also, intramuscular injections into muscles of the rear legs can cause severe pain, lameness, and occasionally peroneal nerve paralysis because of local nerve involvement.

INTRADERMAL INJECTION

Patient Preparation
Intracutaneous (or intradermal) injections are used for diagnostic testing purposes. Prepare the skin by carefully clipping the hair with a No. 40 clipper blade. If the skin surface is dirty, gently clean it with a moist towel. Scrubbing and disinfection are contraindicated because they may produce iatrogenic trauma and inflammation, which interfere with the test.

Technique
Stretch the skin by lifting a fold, and use a 25- to 27-gauge intradermal needle attached to a 1-mL tuberculin syringe. Insert the point of the needle, bevel up, in a forward lifting motion as if to pick up the skin with the needle tip. Advance the needle while pushing the syringe (levered) downward until the bevel is completely within the skin. Inject a bleb of 0.05 to 0.10 mL of fluid. If the procedure is done correctly, the small bleb will appear translucent. Intradermal injections generally are used in patients subjected to intradermal skin testing for allergenic antigens. Administration of compounds by the intradermal technique is not necessarily simple. Inadvertent administration of medications into the subcutaneous tissues is easy when attempting intradermal injection. For that reason, specific training and experience are recommended before attempting intradermal skin testing of allergic patients.

TRANSDERMAL (NEEDLE-FREE) ADMINISTRATION

Patient Preparation
None required.

Technique
Intradermal administration of vaccine and drugs in veterinary and human medicine largely has been limited to the complexities of accurately delivering the desired dose into, and not under, the skin. In 2004 a transdermal administration system* was introduced for cats (recombinant feline leukemia virus [FeLV] vaccine) that was designed after a similar device

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*Vet-Jet Transdermal Administration System, Merial, Duluth, Georgia.
used in human (pediatric) medicine. Recently the transdermal administration system used for administration of the recombinant FeLV vaccine has been re-designed. This same administration system is now used for the transdermal administration of the oral melanoma vaccine. The transdermal administration system consistently delivers a precise volume of vaccine into the skin, subcutaneous tissues, and muscle. Use of the transdermal administration system should only be used to administer those vaccines approved for this method of delivery.

**Special Considerations**

Administration of vaccine using the transdermal administration system requires training to understand proper procedure for loading and administering vaccine. At this writing, sale of the transdermal administration system for delivery of the canine oral melanoma vaccine is limited to select specialists in veterinary medicine.

**Intravenous Injection**

See Section 1.

**Intramuscular Administration**

See Section 1.

**Additional Reading**

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**VASCULAR ACCESS**

See Section 1.

**BANDAGING TECHNIQUES**

See Section 1.

**BLOOD PRESSURE MEASUREMENT: INDIRECT**

**Patient Preparation**

None required.

**Technique**

Generally, two techniques are used. Oscillometric blood pressure (BP) measurement entails use of an automated recording system. A cuff is applied to the base of the tail or a distal limb for access to an artery. This technique generally is regarded as being most accurate in dogs. When oscillometric BP measurements are performed in dogs, the patient should be in lateral recumbency. This places the cuff at approximately the same level as the heart. In cats the patient generally remains in sternal recumbency (and minimally restrained). Most patients experience a brief acclimation period to the cuff placement. For this reason, at least three to five separate readings are obtained at 1- to 2-minute intervals. This technique can be used on awake or anesthetized patients (Figure 4-8).

The Doppler-ultrasonic flow detection system is most accurate in cats for measuring systolic BP. Again, the ventral tail base or a dorsal pedal artery (hindlimb) or the superficial palmar arterial arch (forelimb) can be used. Apply and inflate an occluding cuff. The readings are obtained by a transducer as the pressure on the cuff is reduced. Caution is recommended in interpreting results from dogs that are reported as hypertensive but have no overt clinical disease. The higher reported occurrence of falsely elevated BP in normotensive dogs measured by this method justifies additional scrutiny when interpreting Doppler BP results in dogs.
Clinically, the most common use of indirect BP measurement is in assessing cats for the presence (or absence) of systemic hypertension caused by renal insufficiency or hyperthyroidism (thyrotoxicosis). A common finding among untreated hypertensive cats is retinal detachment and blindness. Early detection and therapeutic intervention (e.g., enalapril and/or amlodipine) is critical. In dogs, BP measurement is indicated in patients with chronic renal insufficiency and/or protein-losing nephropathy, hyperadrenocorticism, and diabetes mellitus. In veterinary medicine, interpretation of BP centers on the systolic BP reading, not the diastolic reading (Table 4-2).

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**DIAGNOSTIC SAMPLE COLLECTION TECHNIQUES**

**Bacterial Culture**

In previous editions of this book, methods of preparing and using selective culture media as well as identifying specific isolates was described. However, technologic advances in microbiology have largely replaced older methods of identifying bacterial isolates in practice. Furthermore, the diverse array of bacterial pathogens, requirements for unique culture media, the risk of sample contamination, and the need for subjective interpretation of
results dictate that even routine bacterial cultures and identification are best reserved for the commercial laboratory equipped to carry out these increasingly complex procedures and experienced in doing so.

What follows are fundamental methods and techniques used to properly collect diagnostic specimens and the appropriate methods for transporting samples to a laboratory in order for the best possible diagnostic result to be obtained.

**Direct Microscopic Examination**

Before actually collecting and submitting a sample to a laboratory for bacterial culture, it is appropriate (whenever feasible to do so) to prepare, stain, and examine, under direct microscopy, exudates or fluid from the suspect material or tissue. Staining the air-dried sample with a rapid Romanowsky-type stain (e.g., Diff-Quik stain) or a Gram stain may reveal evidence of neutrophilic inflammation (neutrophilia, especially with a left shift) and occasionally degenerative neutrophils with intracellular bacteria visible. These findings greatly facilitate patient management by documenting the immediate need for interventive empiric antimicrobial therapy until definitive culture and antimicrobial susceptibility results are obtained. The absence of cytologic evidence of bacterial infection does not rule out the possibility that the patient is infected or bacteremic (Table 4-3).

**Test Considerations**

Collecting diagnostic samples for bacterial culture should be attempted as early in the disease process as possible. It is also critical to accomplish the sample collection under aseptic conditions. It is appropriate, therefore, to perform a surgical scrub of the skin or tissue

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**TABLE 4-3 Common Bacterial Culture Results**

| Site                | Commensals                          | Pathogens                               |
|---------------------|-------------------------------------|-----------------------------------------|
| **EXTERNAL EAR CANAL** |                                     |                                         |
| Dog                 | *Malassezia, Clostridium,*<br>*Staphylococcus* (a few),<br>*Bacillus* (a few); never<br>*Streptococcus,*<br>*Pseudomonas,* or *Proteus* | *Many Staphylococcus and <br>Malassezia together;*<br>*Pseudomonas,* *Proteus,*<br>*Streptococcus,* *Escherichia coli* |
| Cat                 | Not documented                       | *Staphylococcus aureus,*<br>*β-hemolytic streptococci,*<br>*Pasteurella,*<br>*Pseudomonas,* *Proteus,*<br>*E. coli,* *Malassezia* |

**SKIN**

| Site                | Commensals                          | Pathogens                               |
|---------------------|-------------------------------------|-----------------------------------------|
| Dog                 | *Micrococcus, Clostridium,*<br>diphtheroids,*<br>*Staphylococcus epidermidis,*<br>*Corynebacterium,*<br>*Malassezia* | *S. aureus (coagulase positive),* *Proteus,*<br>*Pseudomonas,* *E. coli* |
| Cat                 | *Micrococcus, Streptococcus,*<br>*S. aureus,* *S. epidermidis* | *S. aureus,* *Pasteurella multocida,*<br>*Bacteroides,*<br>*Fusobacterium,*<br>*hemolytic streptococci* |
| Conjunctiva         | *Staphylococcus, Streptococcus,*<br>*Bacillus,*<br>*Corynebacterium,*<br>diphtheroids,*<br>*Neisseria,*<br>*Pseudomonas* | *S. aureus,* *Bacillus,*<br>*Pseudomonas,* *E. coli,* *Aspergillus* |

Continued
from which the sample is to be collected in advance. This is especially true for tissue biopsies and fluid samples collected by needle aspiration through intact skin. Failing to adequately prepare the collection site can result in significant contamination and complicate diagnostic interpretation of results.

In addition, it is recommended to collect the diagnostic sample before the administration of antibiotics in order to minimize the risk of false-negative culture results. In the event antimicrobials have been administered to a patient with a suspected infection, and that is not responding to treatment, discontinuing treatment for 48 hours before attempting sample collection is generally recommended.

Collection of an adequate amount, or volume (fluid), is equally important in obtaining meaningful result. For example, a single sterile cotton-tipped swab of contaminated tissue should be considered inadequate sampling and inappropriate for any patient. Multiple specimens are always recommended when feasible. Also, biopsy material, surgically removed tissue, and several milliliters of fluid (e.g., urine) should be collected and placed in a sterile container that can be appropriately sealed (leak-proof container) before transport. A "clean catch" of urine in a "clean cup" is not appropriate.

Inexpensive commercial containers specifically designed for the transport of infectious material are readily available today and should be used. Many containers designed to hold bacterial samples contain buffered, nonnutritive transport media to sustain the growth of pathogenic bacteria yet minimize overgrowth of bacterial contaminants during the time required to transport the sample. Most commercial laboratories provide appropriate containment devices for the transport of bacterial samples.

**Collection Technique and Sample Transport**

Because most diagnostic specimens collected for bacterial culture are submitted to commercial laboratories for bacterial isolation, identification, and antimicrobial susceptibility testing, it is important to prepare the sample properly for shipping.

Special transport media are generally not required for routine aerobic culture specimens as long as the sample can remain moist and relatively cool and the sample can

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**TABLE 4-3  Common Bacterial Culture Results—Cont'd**

| Site | Commensals | Pathogens |
|------|-------------|-----------|
| Vagina | *Staphylococcus, Streptococcus, Enterococcus, Corynebacterium, E. coli, Haemophilus, Pseudomonas, Peptostreptococcus, Bacteroides* | *Brucella canis; pure culture of organism (especially E. coli, Staphylococcus, Pseudomonas) when accompanied by tissue reaction at vaginal cytology* |
| Urine | <1000* organisms per milliliter; presence of several organisms suggests contamination | More than 100,000* organisms per milliliter and often pure culture; *E. coli, enterobacteria, Klebsiella, Proteus, Pseudomonas aeruginosa, P. multocida, Staphylococcus, Streptococcus* |

*Absolute numbers of bacteria depend on the collection technique.
be inoculated onto culture medium within 3 to 4 hours only. For samples that must be shipped overnight to a laboratory, it is imperative that the specimen be kept cool (not frozen) and moist. Elevated temperatures during shipping contribute to bacterial overgrowth of nonpathogenic bacteria, making isolation and identification of disease-producing organisms difficult. Special transport media may be required. Contact the individual laboratory regarding information pertaining to shipping of specimens for bacterial culture.

Specimens submitted for anaerobic culture need to be inoculated onto culture media within minutes after collection. Although special anaerobic transport media are available, they may not be well suited for extended shipping times (>4 hours).

**Urine**

Among the most frequently tested fluids for bacteria, urine supports the growth of several types of bacteria. Therefore it is necessary that the genitalia be cleaned before collection of urine (free-catch specimen) or cystocentesis (preferred). Use of a urinary catheter to collect urine is likely to introduce urethral bacteria and may result in false-positive culture results. Bacteria will survive for only a limited time in urine. Samples collected must be sealed, and unless processed within 2 hours the sample must be refrigerated. Samples held for longer than 8 hours may not contain viable bacteria. If extended transportation times are required to reach a laboratory, a urine reservation tube (Vacutainer Brand Urine Transportation System, BD, Franklin Lakes, New Jersey) will allow storage for up to 48 hours at room temperature (*Table 4-4*).

**Exudates and Transudates**

Collection from fluid-filled compartments (e.g., abscesses, seromas) requires collection with a needle and syringe. The maximum quantity possible should be collected and submitted. The skin or tissue overlying the area from which the sample is to be collected should be surgically prepared. If it becomes necessary to flush an open lesion (or perform tracheotranstracheal aspiration or bronchoalveolar lavage [BAL]), it is recommended that a buffered solution of sterile Ringer's lactate be used. Use of fluids that contain preservative may actually inhibit the growth of bacteria.

**Feces**

If it is necessary to submit fecal material for specific bacterial isolation, at least 2 to 3 g of feces should be submitted. A single cotton-tipped swab inserted rectally is unlikely to yield meaningful results. Multiple (up to three) samples are recommended when attempting to isolate specific pathogens (e.g., *Salmonella*). Samples should be submitted in a sealed, leak-proof container (always appreciated by the lab). The containerized sample should be refrigerated if there is a significant delay (several hours) involved in submission to the laboratory.

**Blood**

Confirmation of the presence of bacteria in the blood (bacteremia) can be difficult and requires some patient preparation before collection of a series of samples. In addition, samples should be collected only in vials clearly marked for the collection of blood. Furthermore, there are several reasons for an infected patient to have a negative blood culture result—for example, prior or concurrent antimicrobial therapy, chronic (low-grade) infection, and intermittent shedding of bacteria into blood. Sample volume, numbers of samples submitted, skin preparation, and timing of collections are variables that can directly affect results.

Clip and surgically prepare the skin over the cephalic, recurrent tarsal, and/or jugular veins. Do not draw blood for culture through an indwelling intravenous or intraarterial catheter. Collection vials are available for aerobic and anaerobic bacterial culture. It is generally recommended that three blood samples be collected from separate veins over a 24-hour period. There is no advantage to collecting arterial blood. It has been suggested
| Collection Method        | Significant Dogs | Significant Cats | Suspicious Dogs | Suspicious Cats | Contaminant Dogs | Contaminant Cats |
|--------------------------|------------------|------------------|-----------------|----------------|------------------|-----------------|
| Cystocentesis            | ≥1000            | ≥1000            | 100-1000        | 100-1000        | ≤100             | ≤100            |
| Catheterization          | ≥10,000          | ≥1000            | 1000-10,000     | 100-1000        | ≤1000            | ≤100            |
| Voluntary voiding        | ≥100,000†        | ≥10,000          | 10,000-90,000   | 1000-10,000     | ≤10,000          | ≤1000           |
| Manual compression       | ≥100,000†        | ≥10,000          | 10,000-90,000   | 1000-10,000     | ≤10,000          | ≤1000           |

From Osborne CA, Finco DR: *Canine and feline nephrology and urology*, Baltimore, 1995, Williams & Wilkins.

*The data represent generalities. On occasion, bacterial urinary tract infections may be detected in dogs and cats with the fewer organisms (i.e., false-negative results).

†Caution: Because contamination of midstream samples may result in colony counts of 10,000/mL or more in some dogs (i.e., false-positive results), they should not be used for routine diagnostic culture of urine from dogs.
that samples collected during times when the patient is febrile may improve the likelihood of isolating bacteria. The volume of blood collected is determined by the size of the patient, the collection vials (adult, pediatric, infant) used, as well as the laboratory equipment used to propagate the culture. In addition to adult human blood culture collection vials (10 ml), pediatric blood collection vials (5 to 10 mL) and infant collection vials (0.5 to 1.0 mL) are available.

It is appropriate that sterile technique be adhered to during collection of all samples. This includes the use of sterile gloves by the individual collecting the sample. Once blood has been collected, air should not be allowed to enter the collection vial. The vial should be gently inverted (never shaken) two to four times. Vials may be maintained at room temperature (the laboratory maintains samples at 37° C).

The opportunity to submit complementary cultures (e.g., from urine) from patients in which blood cultures are being collected can help to confirm the infecting bacteria and may lead to identification of a likely source (Boxes 4-2 and 4-3).

### Additional Reading

- Dow S: Diagnosis of bacteremia in critically ill dogs and cats. In Bonagura J, editor: Current veterinary therapy XII. Small animal practice, Philadelphia, 1995, WB Saunders.
- Greene CE: Infectious diseases of the dog and cat, ed 3, St Louis, 2006, Elsevier.
- Osborne C: Three steps to effective management of bacterial urinary tract infections, Compend Contin Educ Pract Vet 17:1233–1248, 1995.
- Osborne CA, Finco DR: Canine and feline nephrology and urology, Baltimore, 1997, Williams & Wilkins.
- Scott DW, Miller WH Jr, Griffin CE: Muller and Kirk’s small animal dermatology, ed 5, Philadelphia, 1997, WB Saunders.

### Fungal Culture

Diagnostic fungal cultures depend on selection of the most appropriate culture site and proper collection technique. Fungal cultures are mostly commonly pursued in patients suspected of having superficial fungal infections of the hair, skin, and nails (dermatophytosis).
Samples collected from patients suspected of having fungal infections of the nasal cavity (e.g., aspergillosis) or systemic (also called “deep”) mycotic infections (e.g., histoplasmosis, cryptococcosis) are usually assessed by cytopathology or serology (see Section 5) or with tissue biopsy and histopathology involving special stains.

**Direct Microscopic Examination**

Direct cytologic assessment of samples from patients suspected of having fungal infections is always indicated. You certainly get credit for trying! However, experience in recognizing diagnostic elements of individual fungi and spores is essential, as is the availability of special stains for wet mount (10% potassium hydroxide) cytopathology.

**Collection Technique and Sample Transport**

**Skin and Hair**

Scrapings of skin and plucked hair shafts are commonly selected for fungal culture. The area of skin and hair to be sampled should be cleaned with 70% alcohol. Iodine-based soaps and solutions should not be used. Hair shafts, particularly those immediately adjacent to the lesion, are removed from skin with a sterile hemostat. Skin scrapings can be collected with a sterile surgical blade or the edge of a clean (unused) microscope slide. Scrapings from healthy, normal-appearing skin as well as abnormal skin should be collected. Skin biopsy may be required if results of attempts to culture hair and skin scrapings are negative. Sterile cotton-tipped swabs should not be used to collect samples for fungal culture.

Hair and skin scrapings can be placed directly into a sterile, dry container without need for any type of media as long as the sample can be processed within hours. Refrigeration is generally not required. If transport times are extended, it is reasonable to place samples in a vial containing bacterial transport medium and refrigerate for up to 15 hours. Samples should never be frozen.

**In-Hospital Fungal Culture**

Skin and hair samples from patients suspected of having superficial fungal infections can be inoculated directly on a commercially available substrate called Dermatophyte Test Medium (DTM). Because samples can remain at room temperature and do not require special handling, the use of DTM is ideal for in-hospital use. The medium contains phenol red as a pH indicator. If a dermatophyte is present, characteristic colony morphology will be observed and the medium underlying the colonies will turn red. Vials are unreliable after 2 weeks; color change noted 2 weeks or more after inoculation of the DTM should be disregarded.

**Wood Light**

Ultraviolet light filtered through nickel oxide produces a beam called Wood light. If an animal is taken into a dark room and its hair and skin are exposed to a Wood lamp, fluorescence may occur for several reasons. Hair shafts affected by some species of Microsporum fluoresce a bright yellow-green (like the color of a fluorescing watch face). However, iodide medications, petroleum, soap, dyes, bacteria, and even keratin may produce purple-, blue-, or yellow-colored fluorescence. The positive fungal fluorescence is a valuable aid in selecting affected hairs for culture inoculation. However, a negative fluorescence does not preclude a possible diagnosis of fungal infection. False-negative and false-positive interpretations are common.

**Additional Reading**

Dow S: Diagnosis of bacteremia in critically ill dogs and cats. In Bonagura J, editor: Current veterinary therapy XII. Small animal practice, Philadelphia, 1995, WB Saunders.

Greene CE: Infectious diseases of the dog and cat, ed 3, St Louis, 2006, Elsevier.

Scott DW, Miller WH Jr, Griffin CE: Muller and Kirk's small animal dermatology, ed 5, Philadelphia, 1997, WB Saunders.
**Viral Testing**

**Direct Microscopic Examination**
Microscopic examination of fluid or tissue samples from patients suspected of having a viral infection is unlikely to contribute to the diagnosis. Because viruses are small and generally intracellular particles, neither light microscopy nor viral culture techniques are used in the practice setting. Some commercial and academic laboratories do offer electron microscopy of tissue (cells), fluids, or feces from infected patients, which may allow for direct visualization of virus particles. Results depend on the quality of the sample evaluated, the type of equipment available, and the experience of the individual performing the microscopy.

**Test Considerations**
Various laboratory techniques are currently available for the identification of viral infections in dogs and cats. Excellent qualitative testing platforms are commercially available for in-practice use. Molecular diagnostic tests, viral culture, histopathology, and serology, all of which are routinely available to veterinarians, require that samples be submitted to commercial laboratories for assessment.

**In-Hospital Testing**
Among the in-hospital test systems used to identify virus, the enzyme-linked immunosorbent assay (ELISA) is the most common testing platform used. ELISA testing can be performed quickly (minutes) with little or no patient preparation and with relatively high sensitivity and specificity. Virus (antigen) detection tests are available as point-of-care tests for FeLV antigen in blood or serum and canine parvovirus (CPV) antigen in feces. In addition, these point-of-care tests for viral infections are capable of identifying patients that have not been exposed, enabling the clinician to rule out infection and viral shedding.

Test sensitivity refers to the likelihood that a patient with known infection will have a positive test result (a test with high sensitivity is expected to have few false-positive results).

Test specificity refers to the likelihood that a patient that is free of the infection will have a negative test result (a test with high specificity is expected to have few false-negative results).

In addition, many commercial and point-of-care nonquantitative serologic assays are available that detect antibody to many of the viruses that affect dogs and cats. However, the positive predictive value of antibody tests is typically lower than that of antigen tests. For example, a positive antibody test result to a particular viral pathogen typically does not constitute a diagnosis of infection, especially in the absence of clinical signs. It may merely reflect recent vaccination (e.g., feline immunodeficiency virus). On the other hand, a negative antibody test result generally does indicate that the patient has had no prior exposure to the virus (or vaccine).

**Laboratory-Based Tests**
Serology refers to the use of serum to detect the concentration of antibody and is widely used in veterinary medicine. The value of antibody titers in diagnosing a viral infection is dependent on a number of factors, including the infecting virus, vaccination history, and time since exposure. Use of acute and convalescent antibody titers in a patient suspected of having an acute viral infection can be a reliable diagnostic tool if a fourfold or greater increase in titer can be demonstrated over 2 to 4 weeks. Acute and convalescent viral titers in individual patients are rarely performed in veterinary medicine.

Virus isolation, however, is a valuable diagnostic tool that is underused in veterinary medicine, perhaps because of the limited number of commercial and university laboratories that provide viral isolation services and the increased availability of molecular diagnostic testing services. Diagnosis of viral upper respiratory infection in cats (herpesvirus 1 and/or calicivirus) is perhaps among the situations for which virus isolation can be most useful, especially in cluster households where many shedding carrier cats exist and kittens may be at risk.
To obtain a sample for viral isolation from the oral cavity of a cat, quickly insert a sterile cotton swab into the oral cavity to the level of the tonsil or oropharynx. By rolling the swab across the epithelium, it is possible to harvest cells and virus from infected cats. Immediately place the swab into a virus transport medium (usually provided by the laboratory). Antibiotics added to the solution prevent bacterial overgrowth of the sample. For short-term transit (5 days or less), hold specimens for viral isolation at 4°C rather than frozen. On reaching the laboratory, the specimen will be inoculated into a suitable tissue culture. Within a few days it is usually possible to establish, based on the cytopathic effect on the tissue culture, whether a virus infection is present. Fluorescent antibody testing can be done subsequently to confirm the isolate.

Although availability is limited, direct assessment of specimens (e.g., feces for CPV or canine or feline coronavirus) can be accomplished by electron microscopy. These methods can be useful for infections in which the virus concentration in the specimen reaches $10^6$ to $10^7$ organisms per milliliter. Specimens such as feces, vesicle fluid, brain tissue, urine, or serum can be submitted for electron microscopy.

Tissue specimens and exfoliative cytologic preparations can be submitted for viral identification by histopathology, immunohistochemistry, and direct fluorescent antibody testing. Such testing has limited application in patients with active disease because of the limited availability of these types of services and the time required for samples to be processed and reviewed by a pathologist. These tests can be particularly useful in postmortem diagnostics when multiple animals are potentially at risk.

*Molecular diagnostics* refers to the use of nucleic acid–based tests for the detection of viral DNA or RNA. Polymerase chain reaction (PCR) is a laboratory technology that offers exceptional test sensitivity. Through its ability to amplify trace amounts of DNA or RNA from pathogenic organisms millions of times, PCR facilitates identification of the "target" sequence of nucleic acid and therefore the infecting organism. This technology is also available commercially for the detection of DNA from selected bacteria and rickettsiae. PCR technology is particularly useful in the very early stages of a viral infection, when the level of antibody has not yet reached levels that are detectable with conventional antibody tests. In addition, PCR testing may detect healthy virus carrier animals that pose a risk to a larger population of susceptible animals yet cannot be identified by conventional virus isolation or identification technologies. It should be noted, however, that PCR technology is still subject to false-positive and false-negative test results. Therefore such testing is not necessarily indicated as a primary or exclusive test method for an individual patient.

**Collection Technique and Sample Transport**

**Serology**

Serum, plasma, or other fluids (e.g., cerebrospinal fluid [CSF]) can be tested for the presence of antibodies to selected pathogenic viruses. Whole blood samples should be allowed to thoroughly clot and retract (or the sample should be centrifuged) before serum is collected. Samples are submitted in a leak-proof vial. Refrigeration is appropriate for samples that must be held for several hours before testing.

**Histology and Immunohistochemistry**

Samples are limited to tissue obtained during surgical biopsy. As with conventional histopathology, samples (no more than 5.0 mm thick) should be placed in 10% buffered formalin and submitted in a leak-proof vial. It is recommended that the volume of formalin used be at least 10 times greater than the tissue sample submitted.

**Fluorescent Antibody Testing**

Testing can be performed on tissues collected during surgical biopsy or from tissue impressions (exfoliative) made from tissue imprints on a clean microscope slide. It is recommended that tissue impressions on slides be fixed in alcohol or acetone before submission. Fresh tissue is submitted on wet (not dry) ice and is not subjected to formalin fixation.
Electron Microscopy
Small amounts of tissue suitable for electron microscopy should be no larger than 1 × 2 mm thick. Fixation in 2% to 4% glutaraldehyde for 24 hours at 20°C is required. Feces and body fluids collected for electron microscopy should be submitted fresh, not frozen or fixed in preservative. If shipping is required, feces and body fluids may be refrigerated or shipped on wet ice. Samples should be viable for 48 to 72 hours.

Virus Isolation
Sterile swabs may be used to collect samples for viral culture and isolation. Samples should be inoculated into a sealed vial containing viral transport medium (usually provided by the laboratory). Samples should not be frozen or fixed in preservative.

Polymerase Chain Reaction Testing
Laboratories offering PCR testing typically accept serum or anticoagulated (ethylenediaminetetraacetic acid [EDTA]) whole blood in leak-proof vials. Samples should be refrigerated and shipped on wet ice. Samples should not be frozen.

Blood Collection Techniques
In most instances, a 3- to 5-mL sample of anticoagulated whole blood is adequate for routine hematology; some laboratories will accept as little as 1 mL. For routine biochemical analyses, the volume of serum requested can vary from 1 to 2 mL, depending on the number and type of tests requested. Plan ahead which samples are required to prevent the need for further venipuncture at a later time. In small dogs and cats, using the jugular veins facilitates collection of an adequate volume of blood. If smaller samples are required, the cephalic, lateral saphenous, or medial saphenous vein can be used for sample collection. Do not use the jugular vein if a coagulopathy is suspected, as hemorrhage may be difficult to control after venipuncture.

Patient Preparation
For successful venipuncture, proper restraint of the animal is important. Details for the proper restraint for various venipuncture locations are discussed with each specific topic throughout this text. The patient must remain comfortable yet relatively motionless to avoid iatrogenic vessel laceration. Stretch the skin tightly over the selected vessel without causing vascular occlusion to help anchor the vessel in place during penetration by the needle.

Technique
The specific venipuncture will vary somewhat depending on the specific vein selected. The following sections describe venipuncture technique for each of four commonly accessed veins: the cephalic vein, jugular vein, lateral saphenous vein, and medial saphenous vein.

Cephalic Venipuncture
To restrain a dog or cat for venipuncture of the cephalic vein, place the dog or cat on the table, sitting or in sternal recumbency. If the right vein is to be tapped or catheterized, the assistant should stand on the left side of the animal and place the left arm or hand under the animal’s chin to immobilize the head and neck. The assistant should reach across the animal and grasp the leg just behind and distal to the right elbow joint. The assistant should use the thumb to occlude and rotate the cephalic vein laterally while the palm of the hand holds the elbow in an immobilized and extended position. Make sure that the animal stays on the table if struggling occurs. The person performing the venipuncture then grasps the leg at the metacarpal region and begins the venipuncture on the medial aspect of the leg, just adjacent to the cephalic vein proximal to the carpus.

Jugular Venipuncture
For a jugular venipuncture in the dog, place the patient in sternal recumbency, with the hands of the assistant placed around the patient’s muzzle to extend the neck and nose dorsally toward the ceiling. In short-coated dogs, the jugular vein usually can be seen coursing
from the ramus of the mandible to the thoracic inlet in the jugular furrow. The vessel may be more difficult to visualize in dogs with long hair coats or if excessive subcutaneous fat or skin is present. The person performing the venipuncture should place the thumb of the nondominant hand across the jugular vein in the thoracic inlet or proximal to the thoracic inlet to occlude venous drainage from the vessel and allow it to fill. With the dominant hand, the person performing the venipuncture should insert the needle and syringe or Vacutainer (BD, Franklin Lakes, New Jersey) into the vessel at a 15- to 30-degree angle to perform the venipuncture.

For smaller and very large animals, the jugular vein also can be tapped by placing the patient in lateral recumbency. The assistant should pull the animal's front legs caudally and extend the head and neck so that the jugular vein can be visualized. The venipuncture then can be performed as previously described. A jugular venipuncture is contraindicated in patients with thrombocytopenia or vitamin K–antagonist rodenticide intoxication.

Place cats in sternal recumbency. The assistant should stand behind the patient so that the patient cannot back away from the needle during the venipuncture. The assistant should extend the cat's head and neck dorsally while restraining the cat's front legs with the other hand. The cat's fur can be clipped or moistened with isopropyl alcohol to aid in visualization of the jugular vein as it stands up in the jugular furrow. The person performing the venipuncture should occlude the vessel at the thoracic inlet and insert the needle or Vacutainer apparatus into the vessel as previously described to withdraw the blood sample. Alternately, place the cat in lateral recumbency as described in the previous paragraph.

**Lateral Saphenous Venipuncture**

To perform a lateral saphenous venipuncture, place the patient in lateral recumbency. The lateral saphenous vein can be visualized on the lateral portion of the stifle, just proximal to the tarsus. The assistant should extend the hindlimb and occlude the lateral saphenous vein just proximal and caudal to the tarsus. The person performing the venipuncture should grasp the distal portion of the patient's limb with the nondominant hand and insert the needle or Vacutainer apparatus with the dominant hand to withdraw the blood sample.

**Medial Saphenous Venipuncture**

To perform a medial saphenous venipuncture, place the patient in lateral recumbency. Move the top hindlimb cranially or caudally to allow visualization of the medial saphenous vein on the medial aspect of the tibia and fibula. The assistant should scruff the patient, if the patient is small, or should place the forearm over the patient's neck to prevent the patient from getting up during the procedure. With the other hand, the assistant should occlude the medial saphenous vein in the inguinal region. The person performing the medial saphenous venipuncture should grasp the paw or hock of the limb and pull the skin taut to prevent the vessel from rolling away from the needle. The fur may be clipped or moistened with isopropyl alcohol to aid in visualization of the vessel. The needle or Vacutainer apparatus can be inserted into the vessel at a 15- to 30-degree angle to withdraw the blood sample.

**Special Considerations**

Incorrect proportions of blood to anticoagulant may result in water shifts between plasma and red blood cells (RBCs). Such shifts may alter the packed cell volume (PCV), especially when small amounts of blood are added to tubes prepared with volumes of anticoagulant sufficient for much larger volumes of blood. Erroneous laboratory results also may be obtained when small volumes of blood are placed in a relatively large container. Evaporation of plasma water and adherence of the cells to the surface of the container can produce artifactual changes in hematologic results.

Refrigerate liquid blood mixed with anticoagulant after collection if the sample is to be held before being transported to a laboratory. White blood cell (WBC) and RBC counts, PCV, and hemoglobin level can be measured within 24 hours of sample
collection. Platelet counts, however, should be done within 1 hour of collection. Dried, unfixed blood smears can be stained with most conventional stains 24 to 48 hours after being made. If a considerable delay is anticipated between the time that the blood smear is made and the staining process, the blood smear should be fixed by immersion in absolute methanol for at least 5 minutes. Blood smears fixed by this method are stable indefinitely.

Never place unfixed blood smears in a refrigerator because condensation forming after the smear is removed from the refrigerator will ruin the blood smear and make it unusable for cytologic evaluation. Take care to leave unfixed blood smears face down on a countertop or in a closed box. Special stains, such as peroxidase, may require fresh blood films.

**Routine Hematologic Testing (See Also Section 5)**

The anticoagulant of choice for hematologic testing is EDTA. Heparin is especially to be avoided if blood films are to be made from blood mixed with anticoagulant because contact with whole blood will distort the morphology of cells significantly. Heparin is acceptable for most procedures requiring blood plasma. The anticoagulant effect of heparin is transitory. Specimens still may clot after 2 to 3 days.

Make blood films immediately after collection because cell morphology rapidly deteriorates after sample collection. Although blood films can be made after introducing blood to EDTA, a better practice is to make blood smears (films) immediately from the collection needle before the blood comes in contact with any anticoagulant. Never use blood exposed to heparin to make blood smears.

Incorrect proportions of blood to anticoagulant may result in water shifts between plasma and RBCs. Such shifts may alter the PCV, especially when small amounts of blood are added to tubes prepared with volumes of anticoagulant sufficient for much larger volumes of blood. Erroneous laboratory results also may be obtained when small volumes of blood are placed in a relatively large container. Evaporation of plasma water and adherence of the cells to the surface of the container can produce artifactual changes in hematologic results.

Refrigerate liquid blood mixed with anticoagulant after collection if there is a delay in making the laboratory determinations. WBC and RBC counts, PCV, and hemoglobin level can be measured within 24 hours of sample collection. Platelet counts, however, should be done within 1 hour of collection. Dried, unfixed blood smears can be stained with most conventional stains 24 to 48 hours after being made. If a considerable delay is anticipated between the time that the blood smear is made and the staining process, the blood smear should be fixed by immersion in absolute methanol for at least 5 minutes. Blood smears fixed by this method are stable indefinitely. Never place unfixed blood smears in a refrigerator because condensation forming after the smear is removed from the refrigerator will ruin the blood smear and make it unusable for cytologic evaluation. Take care to leave unfixed blood smears face down on a countertop or in a closed box. Special stains, such as peroxidase, may require fresh blood films.

**Routine Biochemistry Testing (See Also Section 5)**

**Patient Preparation**

Prepare the selected vein as described earlier.

**Technique**

Most clinical chemistry procedures are performed on serum. The serum is obtained by collecting blood without any anticoagulant and allowing the blood to clot in a clean, dry tube. Separate serum from cells within 45 minutes of sample collection (venipuncture). Special vacuum vials are available that produce a gel barrier between the clot and the serum (serum separator tubes) which avoid the need to draw off the serum into a separate vial. Clotting of the blood and retraction of the clot occur best and maximum yields of serum are obtained at room or body temperature. Refrigeration of the sample delays clot retraction. Some samples clot and retract faster than others.
**Special Considerations**

If a serum separator tube is not used, it is recommended to free the clot from the walls of the container by rimming with an applicator stick. After the clot is freed, allow clot retraction to occur, and then centrifuge and draw off the clear supernatant serum using a pipette or suction bulb. Allow whole blood samples to completely clot before attempting to remove serum. Failing to do so may result in a mixture of plasma and serum in the submitted sample. Serum yield is usually one third of the whole blood volume. Patients that are hypovolemic or dehydrated can have a significantly lower serum yield.

Many clinical chemistry procedures can be performed on plasma and on serum. The advantage of using plasma is that separation of cells can be accomplished immediately after centrifugation or sedimentation, without the need to wait for clot formation and retraction. The disadvantage of plasma is that the presence of the anticoagulant interferes with many of the chemistry assay procedures. Plasma is less clear than serum, which may be an additional disadvantage for colorimetric assays. Plasma and serum are virtually identical in chemical composition except that plasma has fibrinogen and the anticoagulant. For many procedures in which plasma or whole blood is to be used, heparin is the anticoagulant of choice. Heparinized blood is the only acceptable specimen for blood pH and blood gas analyses. Although blood containing EDTA is acceptable for certain chemical procedures, it cannot be used for determination of plasma electrolytes because it contributes to and sequesters them from the specimen. In addition, EDTA can interfere with alkaline phosphatase levels, decrease total carbon dioxide, and elevate blood nonprotein nitrogen.

Refer to the Tube Selection Guide in Section 5 to assure use the proper collection tube is used for the appropriate test requested.

Separate serum or plasma and remove it from the cells as soon as possible after blood is collected, because many of the constituents of plasma exist in higher concentrations in RBCs. With time, these substances leak into the plasma and cause falsely elevated values (positive interference) and falsely lower values (negative interference) (Table 4-5). Under no circumstances should whole blood be sent via the mail; serum derived from such specimens usually is hemolyzed, and results are often inaccurate. Separate serum and transfer it to a clean, dry tube for shipment.

| **Table 4-5** Examples of Positive and Negative Interference on Biochemistry Analytes Induced by Sample Hemolysis |
|-----------------|------------------|
| **Analyte**     | **Effect of Hemolysis*** |
| Alanine transaminase | Minimal effect |
| Alkaline phosphatase | Increased |
| Bilirubin        | Increased |
| Chloride         | Decreased |
| Creatinine       | Increased |
| Inorganic phosphate | Increased |
| Lipase           | Decreased |
| pH               | Decreased |
| Potassium        | No detectable effect |
| Total calcium    | Increased |
| Total protein    | Increased |
| Urea nitrogen    | Increased |

*Type and degree of interference vary among different testing modalities unique to individual laboratories or in-hospital biochemistry analyzers.
BONE MARROW ASPIRATION

Collection of bone marrow may prove valuable in diseases of the blood in which examination of the peripheral blood reveals abnormal cells or cell counts. Conditions such as leukopenia, thrombocytopenia, nonregenerative anemia, agranulocytosis, pancytopenia, leukemias, other bone marrow cancers, and infectious diseases (e.g., histoplasmosis,ehrlichiosis) may be confirmed only by assessment of bone marrow cytology.

Bone marrow in the young animal is cellular and exists in the flat bones (sternum, ribs, pelvic bones, and vertebrae) and in the long bones (humerus and femur). As the animal ages, the cellular content of the marrow decreases, especially in the long bones. In older animals, bone marrow cells still exist in the flat bones; however, in conditions of stress in which new blood cells must be produced in large numbers, primitive cells in the bone marrow of the long bones again become active. Interpretation of the bone marrow smear may be limited by (1) technique used to obtain a bone marrow specimen or (2) the specialized knowledge necessary to interpret bone marrow cells.

Bone marrow aspiration is much underused in clinical practice. The procedure does require some degree of skill if high-quality samples are to be obtained, but the procedure is of low risk to the patient and can be highly valuable in establishing a diagnosis or prognosis.

Canine

Patient Preparation

A short-acting anesthetic occasionally may be needed, but tranquilization together with infusion of local anesthetic is usually sufficient. The site selected for aspiration or biopsy must be shaved and surgically prepared.

Technique

Bone marrow aspiration or biopsy is a percutaneous procedure conducted using sterile technique.

The techniques involved include marrow aspiration and bone marrow core biopsy alone or in combination. When aspiration biopsy fails to produce adequate cytology (as in advanced myelofibrosis, neoplasia, or marrow aplasia), a core biopsy of bone marrow is indicated. The bone marrow aspiration needle may be a 16-gauge Rosenthal needle or Illinois needle for a medium-sized dog; an 18-gauge Rosenthal needle for a small dog or a cat; or a Jamshidi (pronounced yam-she-dee) bone marrow biopsy needle, 12 gauge for most adult dogs and 14 gauge for small dogs and cats.

The selection of needles for aspiration biopsy of bone marrow is based on the biopsy site, the depth of the biopsy site, and the density of cortical bone. For bone marrow aspiration, the modified disposable Illinois sternal-iliac bone marrow aspiration needle works well (Figure 4-9). For a core biopsy of bone marrow, the Jamshidi bone marrow biopsy-aspiration needle (pediatric, 3.5 inches, 13 gauge) can be used (Figure 4-10).

The iliac crest is a commonly used site for marrow aspiration in dogs. Place the animal in lateral recumbency, and prepare the aspiration site. To aspirate marrow, have the needle enter the widest part of the iliac crest and stop the needle just after penetration of the bone. Remove the stylet, place a 12-mL syringe on the needle, and aspirate 0.2 mL of marrow.

Alternatively, the head of the humerus offers easy access to abundant bone marrow. Sedation may be required. With the patient in lateral recumbency and the humerus flexed (the humerus is positioned parallel to the patient’s thorax), instill local anesthetic into the
skin and subcutaneous tissues to the level of the head of the humerus. The site of needle insertion is on the most proximal facet of the humeral head (Figure 4-1). Direct the needle into the bone toward the elbow and parallel to the humeral shaft. If the needle is positioned too far medially over the humeral head, it is easy to penetrate the joint capsule. Although this is a common occurrence, it does not pose a risk of injury to the patient (assuming the skin was surgically prepared). If joint fluid contaminates the bone marrow aspirate, the sample will be rendered useless.

Contamination of the bone marrow with peripheral blood results if (1) the marrow is not aspirated immediately after the needle enters the marrow cavity or (2) if aspiration time is sustained and a large volume of blood enters the syringe subsequent to the rupture of small blood vessels in the bone marrow.

Figure 4-9: Illinois iliac-sternal bone marrow needle used for aspiration of bone marrow from the humerus, ileum, or femur of dogs and cats.

Figure 4-10: Jamshidi bone marrow biopsy needles.
Perhaps the least desired technique is to obtain marrow from the proximal end of the femur by insertion of the bone marrow needle into the trochanteric fossa. Make a small skin incision over the trochanteric fossa just medial to the summit of the trochanter major. Insert the bone marrow aspiration needle medial to the trochanter major, and place the long axis of the needle parallel to the long axis of the femur.

Once the site has been selected, grasp the needle firmly. Apply steady, slight pressure while alternately rotating the needle tip against the bone (fast, 180-degree clockwise and then counterclockwise movements). Begin with gentle pressure until the needle begins to seat into the bone. Gradually increase the pressure as the needle penetrates into the bone. Insert the bone marrow needle ½ inch into the femoral canal. Remove the stylet from the needle, and aspirate using a 12- or 20-mL syringe that contains a small volume (approximately 0.1 mL) of 4% EDTA. Use significant negative pressure, for example, by withdrawing the plunger of a 12-mL syringe to the 8- or 9-mL mark. Collection of more than 1 mL of bone marrow is unnecessary. Collection of larger volumes may cause greater amounts of peripheral blood to enter the syringe, leading to hemodilution of the sample. Once collection is complete, immediately transfer the aspirate to a watch glass containing approximately 0.25 mL of 4% EDTA. Immediately mix the sample well using the end of the syringe. This is also a good time to remove the bone marrow needle from the patient.

Prepare slides in a manner similar to that used for peripheral blood smears. Preparation of five to eight high-quality slides for submission is customary. Smears are air-dried. Slides may be stained using the same stains used for peripheral blood smears.

Bone marrow biopsy samples, usually obtained as a core, should be placed directly into 10% buffered formalin. It is generally recommended not to roll the core across a microscope slide (exfoliative cytology), as this may significantly disrupt the architecture of the sample and influence histopathologic interpretation.

Special Considerations

When submitting a bone marrow aspirate or bone biopsy, a complete blood count (CBC) should also be collected from that patient on the same day. The bone marrow sample and the CBC should be submitted together in order to obtain maximum diagnostic information. A thorough patient history should accompany the submitted samples.

Depending on the volume of bone marrow aspirate obtained, any additional aspirated bone marrow remaining after slides have been made can be mixed with EDTA in the same
type of tube used to collect whole blood for a CBC. Tubes may be refrigerated for short periods but never frozen. Prompt shipping and processing of liquid samples of bone marrow is encouraged, as these cells tend to rapidly undergo degeneration.

Bone marrow biopsy core samples, after fixation in 10% buffered formalin, require decalcification before processing and interpretation.

Feline

**Patient Preparation**

When feasible, a short-acting anesthetic administered to a cat before bone marrow aspiration or biopsy is recommended owing to the difficulty of adequately restraining a cat, even if sedated. The site selected for aspiration or biopsy must be shaved and surgically prepared. Infusion of local anesthetic at the aspiration or biopsy site is appropriate. Supplemental oxygen may be indicated.

**Technique**

Accessible sites for bone marrow sampling and biopsy in the cat are the iliac crest, the head of the humerus, and the proximal end of the femur via the trochanteric fossa. The techniques described for the dog can be used.

Smears of bone marrow are made immediately after aspiration. Extrinsic thromboplastin present in bone marrow tissue will cause the marrow to clot within 30 seconds. Unstained slides should be submitted. A core of bone marrow can be fixed in 10% buffered formalin before submission for decalcification and histologic preparation.

Another method is to aspirate the sample of bone marrow into a syringe containing 0.25 mL of 4% EDTA solution. Expel the aspirate, up to 0.5 mL, into a sterile Petri dish, from which the marrow particles can be isolated easily by aspirating an aliquot with a glass pipette, placing an appropriate volume onto several glass slides, making the appropriate number of smears.

**Special Considerations**

Slides prepared from bone marrow aspirates should be allowed to air-dry and then labeled appropriately. Slides should never be refrigerated, as moisture from condensation can alter or destroy the appearance of individual cells.

**Additional Reading**

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**Cytology Collection Techniques**

(See also Section 5 for additional information on slide preparation of samples to be submitted for cytopathologic examination.)

Cytopathology involves a simple, direct, and inexpensive technique that can yield significant diagnostic information within a short time at minimal direct cost. Cytologic examination can be made of material obtained from pustules, vesicles, or the raw, ulcerated, or cut surfaces of a lesion. To make the smear, press a clean microscope slide firmly against a raw or ulcerated lesion to transfer cellular material to the slide. Exudates may be collected by sterile swab or may be aspirated into a sterile syringe. Roll the swab gently across the slide, or place a drop of fluid from the syringe onto the slide and carefully spread the fluid in a uniform film. Transfer material from a block of tissue to the slide by gently pressing the tissue onto the slide in several locations. Use various stains for different conditions.

Rapid stains such as new methylene blue or a quick Romanowsky-type stain (e.g., Diff-Quik) are useful and convenient for office procedures. Even Wright and Gram stains for evaluation of bacteria in tissues and fluids are easy to use. The presence of many bacteria,
especially mixed types, may mean only surface contamination, whereas single types of bacteria, abundant polymorphonuclear WBCs, and especially phagocytosis support the diagnosis of infection and the host response to it. A few acantholytic cells (loose epidermal cells) in the smear may be compatible with infectious processes, but large numbers, or “rafts,” of acantholytic cells are highly suggestive of pemphigus and imply the need for more complex tests for positive diagnosis.

Large numbers of eosinophils sometimes are found in stained smears. Contrary to popular opinion, they usually do not mean allergy. These cells are seen most commonly with furunculosis and may be associated with the eosinophilic granulomas, eosinophilic plaques, sterile eosinophilic pustulosis, pemphigus complex, and ectoparasites. Yeasts (usually Malassezia, rarely Candida) commonly are found as budding cells in masses of wax and debris from ear smears.

Tumor cells may be recognized in some impression or aspiration samples where Giemsa is a preferred stain. Although special expertise is needed, cases of mastocytoma, histiocytoma, and lymphoma are recognized most easily. Always prepare formalin-fixed tissues for histologic diagnosis in tumor evaluations (Box 4-4).

**Percutaneous Fine-Needle Aspiration**

**Patient Preparation**

Fine-needle aspiration, the use of needle and syringe to remove cells from normal and abnormal tissue, apply them to a glass slide, stain the smear, and review the results immediately is among the most useful, cost-effective procedures available in clinical practice. In most cases there will be no specific requirements for patient preparation. Shaving hair over the aspiration site is generally not required. Surgical preparation of the site is optional.

**Technique**

Lymph node aspiration is a procedure that can, and should, be performed routinely in clinical practice. Follow proper technique to maximize the diagnostic utility of this procedure. Lymph node aspiration typically is indicated (1) in patients with generalized lymphadenomegaly, (2) to evaluate abnormally enlarged solitary lymph nodes, and (3) in suspected instances of tumor metastases to lymph nodes. Surgically prepare the skin over the node from which a biopsy specimen is to be taken. With one hand, localize and immobilize the lymph node; with the other hand, guide the aspiration biopsy needle into the affected node. Affix a 6-mL syringe onto a 22- to 20-gauge needle (a 25-gauge needle can be used when the site to be aspirated is particularly small), and advance the needle into the lymph node. Withdrawal of the syringe to approximately 0.5 mL before inserting it into the tissue is recommended. Doing so helps to prevent expelling material when removing the sample from the tissue. When the needle is in position in the approximate center of the node, gradually draw negative pressure on the syringe to a level of 4 to 5 mL. Hold the negative pressure in place for a few seconds. Release, and then repeat two or three times. Before removing the needle from the tissue, release the negative pressure in the syringe (this is why it is recommended to have 0.25 mL of air prepositioned inside). Do not remove the syringe from the tissue while maintaining negative pressure, because this can

| **BOX 4-4** CYTOLOGIC FEATURES OF MALIGNANCY |
|---------------------------------------------|
| Enlargement of nucleus or nuclei larger than 10 nm | Increase in size and number of nucleoli |
| Decreased nuclear/cytoplasmic ratio | Increased basophilia of cellular cytoplasm; increased RNA content |
| Multinucleation because of abnormal mitosis | Anisokaryosis or pleomorphism |
| Abnormal or frequent mitosis | Multinucleated giant cells |
| Variations in size and shape of nuclei |                                     |
result in the aspiration of significant amounts of blood from the skin, thereby significantly diluting the sample with peripheral blood. Eject cellular material within the needle onto clean glass slides. Handle all aspirates gently. To make slides, place two slides together and pull the slides apart to avoid shearing the cells. Do not compress or force slides together. In addition, a biopsy of the lymph node can (and usually should) be performed as a means of confirming or supporting diagnostic decisions made on aspirates. Lymph node biopsy samples can be obtained easily and safely by punch (core) techniques (e.g., 4-mm skin biopsy punch) or Tru-Cut biopsy needle.

Special Considerations
The most significant limiting factors are (1) the technical ability to prepare high-quality slides and (2) the ability to interpret the cytologic findings. Some experience is needed to obtain the skills needed to aspirate cells and make diagnostic preparations. Significant training is required to interpret the slides adequately. However, access to cytopathologists affiliated with diagnostic laboratories today makes fine-needle aspiration a highly useful diagnostic tool. The lymph node aspiration technique, described next, illustrates the finer points of the fine-needle aspiration technique.

Exfoliative Cytology
Patient Preparation
Also called “touch impression cytology,” exfoliative cytology entails preparing cytologic slides directly from the cut surface of biopsy samples. Requirements for patient preparation depend on the location of the tissue from which the sample is taken. The number and quality of cells collected are best when the procedure involves the freshly cut surface of tissue. Attempting to collect samples directly from skin lesions on the patient is much less likely to yield diagnostic cytology. Preparation, therefore, depends on the target tissue from which slides are needed. Preparation may entail local anesthesia and collection of a tissue biopsy specimen from a lesion or suspect tissue (e.g., lymph node or cutaneous tumor) or general anesthesia and an exploratory abdominal procedure (e.g., liver biopsy).

Technique
Once the tissue has been collected, a scalpel blade is used to make a full-thickness linear cut through the biopsy specimen. A fresh surface of the tissue of interest is exposed. Using forceps or a sterile needle, gently lay the tissue on a clean glass slide. Do not force the tissue onto the slide, because this can significantly damage cells. Several imprints can be made from the same surface. As needed, make new cuts to obtain a fresh surface from which to exfoliate cells. Allow the slide to air-dry completely. Apply conventional staining, and examine the specimen when it is dry. The remaining tissue, if not significantly damaged, can be submitted for histopathologic examination (recommended).

Special Considerations
Once slides have air-dried and are labeled, they may be stained and reviewed immediately or submitted for review and interpretation by a pathologist. Unstained slides should not be refrigerated, as moisture from condensation can alter the cytology of the preparation. Several slides should be submitted. Any remaining tissue may be placed in 10% buffered formalin and submitted for histopathology.

The number of diagnostic cells obtained when making slides by way of exfoliative cytology depends on the tissue. Epithelial cells (e.g., carcinoma), mast cells (cutaneous mast cell tumor), and lymph nodes readily exfoliate abundant numbers of cells. Excessively thick slides can make interpretation difficult. On the other hand, biopsy specimens of tissue composed predominantly of mesenchymal cells (e.g., granuloma, fibrosarcoma) do not readily exfoliate cells, and slides made from these types of tissues are typically hypocellular.
Contamination of the cytology specimen with peripheral blood is a common mistake that can make interpretation of the sample difficult. It may be appropriate to gently blot the cut surface of the tissue sample, thereby removing excessive blood, before making slides.

**Scrapings and Swabs**

Depending on the tissue type and lesion, it may be possible to obtain diagnostic cytologic samples from scrapings (e.g., conjunctival epithelium for virus inclusions), brushes (e.g., material obtained during endoscopy), and swabs (e.g., ear and vaginal swabs). The cells, once harvested, can be applied delicately directly to a clean glass slide by carefully rolling or even by just touching the material to the slide to create a thin layer. Allow the sample to air-dry thoroughly before staining.

**Fluids**

Cytologic examination of fluids obtained with needle and syringe from body cavities, cysts, and urine typically requires additional preparation to obtain adequate cell concentration to make diagnostic decisions. Analyze fluid specimens with respect to protein and nucleated cell count and a morphologic description of the cells. If overall cell counts are low, centrifugation will be required to concentrate cellular material for analysis. After centrifugation, remove the supernatant (and save it). Resuspend the cells in two or three drops of the supernatant. Apply a single drop of the mixture to a glass slide and allow it to air-dry. I prefer not to smear the liquid onto the slide; instead, I allow the liquid to run, by gravity, from one end of the slide to the other. After the liquid is thoroughly air-dried, it can be stained and reviewed.

**Ectoparasites**

**Skin Scrapping**

**Patient Preparation**

None required.

**Technique**

Skin scrapings frequently are obtained to find and identify microscopic parasites or fungal elements in the skin. Material required includes mineral oil in a small dropper bottle, a dull scalpel blade, glass slides, coverslips, and a microscope.

Select undisturbed, untreated skin for a scraping site. The best method is to scrape the periphery of skin lesions and avoid the excoriated or traumatized center areas. In scraping for demodectic mange, pinch a small fold of affected skin firmly and collect the surface material for examination. This procedure forces the mites out of the hair follicles and onto or near the skin surface. For sarcoptic mange, scrape large areas. Select sites on the elbows, hocks, and ear margins when searching for sarcoptic mange. Many or frequent scrapings may be necessary to demonstrate sarcoptic mange mites or their fecal pellets or eggs.

Place the accumulated material on a microscope slide and mix it with mineral oil. Examine the entire area with a ×10 objective thoroughly and carefully.

**Acetate Tape Preparation**

**Patient Preparation**

None required.

**Technique**

Acetate tape preparation is one of the simplest diagnostic procedures to perform when looking for the presence of ectoparasites, especially the nits of *Cheyletiella*. Use clear (not frosted) acetate tape. Bend the tape into a loop around the fingers with the sticky side facing out. Part the animal’s hair coat, and press the tape firmly onto the skin and hair around suspect lesions. The sticky tape picks up loose particles with which it makes contact. Cut the loop of tape and place the strip of tape sticky side down on a clean microscope slide. Use a
low-power microscope to look through the tape at the collected particles. This technique is excellent for trapping and identifying biting and sucking lice, *Otodectes* and *Cheyletiella* mites, flea dirt and larvae, fly larvae, and dandruff scales.

Acetate tape also is useful for studying hair abnormalities. Use a strong hemostat to securely clamp and quickly avulse a group of 10 to 20 hair shafts. Press the pointed distal ends onto sticky acetate tape (lined up like pickets in a fence), and cut the hair shafts off in the middle with scissors. Likewise, press the butt ends with the hair roots onto another piece of tape. Then press the tape holding the hair onto a microscope slide to allow low-power examination of the hairs through the clear tape. The tips of the hairs will be well oriented and controlled; thus, it is easy to evaluate whether the hairs are split, broken, or bitten off and whether the hair roots are in the anagen or telogen growth stage.

**Additional Reading**

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Ehrhart N: Principles of tumor biopsy, *Clin Tech Small Anim Pract* 13:1998.

**Urine Collection Techniques**

Urine can be removed from the bladder by one of four methods: (1) voided (the “free catch”), (2) manual compression of the urinary bladder (expressing the bladder), (3) catheterization, or (4) cystocentesis.

**Voiding**

For routine urinalysis, collection of urine by voiding (micturition) is satisfactory. The major disadvantage is risk of contamination of the sample with cells, bacteria, and other debris located in the genital tract and the perineal hair coat. The first portion of the stream is discarded, as it is most likely to contain debris. Voided urine samples are not recommended when bacterial cystitis is suspected.

**Manual Compression of the Bladder**

Compressing the urinary bladder is occasionally used to collect urine samples from dogs and cats. *Critical: Do not use excessive pressure*; if moderate digital pressure does not induce micturition, discontinue the technique. Excessive pressure can culminate in forcing contaminated urine (bladder) into the kidneys, or, worse, in patients with a urethral obstruction the urinary bladder can rupture. The technique is most difficult to accomplish in male dogs and male cats.

**Urinary Catheterization**

Several types of urinary catheters are currently available for use in dogs and cats. The catheter types most often used today are made of rubber, polypropylene, and latex-free silicone. Stainless steel catheters are occasionally used but unless placed with care these can cause damage to the urethra and/or urinary bladder. Generally, urinary catheters serve one of four purposes:

1. To relieve urinary retention
2. To test for residual urine
3. To obtain urine directly from the bladder for diagnostic purposes
4. To perform bladder lavage and instillation of medication or contrast material

The size of catheters (diameter) usually is calibrated in the French scale; each French unit is equivalent to roughly 0.33 mm. The openings adjacent to the catheter tips are called “eyes.” Human urethral catheters are used routinely in male and female dogs; 4F to 10F catheters are satisfactory for most dogs (*Table 4-6*). Polypropylene catheters should be individually packaged and sterilized by ethylene oxide gas.
Catheterization of the Male Dog

Patient Preparation

Equipment needed to catheterize a male dog includes a sterile catheter (4F to 10F, 18 inches long, with one end adapted to fit a syringe), sterile lubricating jelly, povidone-iodine soap or chlorhexidine, sterile rubber gloves or a sterile hemostat, a 20-mL sterile syringe, and an appropriate receptacle for the collection of urine.

Proper catheterization of the male dog requires two persons. Place the dog in lateral recumbency on either side. Pull the rear leg that is on top forward, and then flex it (Figure 4-12). Alternatively, long-legged dogs can be catheterized easily in a standing position.

Before catheter placement, retract the sheath of the penis and cleanse the glans penis with a solution of povidone-iodine 1% or chlorhexidine. Lubricate the distal 2 to 3 cm of the appropriate-size catheter with sterile lubricating jelly. Never entirely remove the catheter from its container while it is being passed because the container enables one to hold the catheter without contaminating it.

Technique

The catheter may be passed with sterile gloved hands or by using a sterile hemostat to grasp the catheter and pass it into the urethra. Alternatively, cut a 2-inch “butterfly” section from the end of the thin plastic catheter container. This section can be used as a cover for the sterile catheter, and the clinician can use the cover to grasp and advance the catheter without using gloves.

If the catheter cannot be passed into the bladder, the tip of the catheter may be caught in a mucosal fold of the urethra or there may be a stricture or block in the urethra. In small-breed dogs, the size of the groove in the os penis may limit the size of the catheter that can be passed. One also may experience difficulty in passing the catheter through the urethra where the urethra curves around the ischial arch. Occasionally a catheter of small diameter may kink and bend on being passed into the urethra. When the catheter cannot be passed

| TABLE 4-6 | Recommended Urethral Catheter Sizes for Routine Use in Dogs and Cats |
|-----------------|-----------------------------------------------------|
| **Animal**       | **Urethral Catheter Type**                                      |
| Cat              | Flexible vinyl, red rubber, or Tom Cat catheter (polyethylene) | 3.5 |
| Male dog (≤25 lb) | Flexible vinyl, red rubber, or polyethylene                  | 3.5 or 5 |
| Male dog (≥25 lb) | Flexible vinyl, red rubber, or polyethylene                  | 8 |
| Male dog (>75 lb) | Flexible vinyl, red rubber, or polyethylene                  | 10 or 12 |
| Female dog (≤10 lb) | Flexible vinyl, red rubber, or polyethylene                  | 5 |
| Female dog (10-50 lb) | Flexible vinyl, red rubber, or polyethylene                  | 8 |
| Female dog (>50 lb) | Flexible vinyl, red rubber, or polyethylene                  | 10, 12, or 14 |

From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat and rabbit, ed 2, Philadelphia, Lippincott-Raven, 1997.

*The diameter of urinary catheters is measured on the French (F) scale. One French unit equals roughly 0.33 mm.
on the first try, reevaluate the size of the catheter and gently rotate the catheter while passing it a second time. Never force the catheter through the urethral orifice.

**Special Considerations**
Effective catheterization is indicated by the flow of urine at the end of the catheter, and a sterile 20-mL syringe is used to aspirate the urine from the bladder. Walk the dog immediately after catheterization to encourage urination.

**Catheterization of the Female Dog**

*Patient Preparation*
Equipment needed to catheterize a female dog includes flexible urethral catheters identical to those used in the male dog. The following materials also should be on hand: a small nasal speculum, a 20-mL sterile syringe, lidocaine 0.5%, sterile lubricating jelly, a focal source of light, appropriate receptacles for urine collection, and 5 mL of povidone-iodine or a dilute chlorhexidine solution.

Use strict asepsis. Cleanse the vulva with a solution of povidone-iodine or dilute chlorhexidine. Instillation of lidocaine 0.5% into the vaginal vault helps to relieve the discomfort of catheterization. The external urethral orifice is 3 to 5 cm cranial to the ventral commissure of the vulva. In many instances the female dog may be catheterized in the standing position by passing the female catheter into the vaginal vault, despite the fact that the urethral papilla is not visualized directly.

*Technique*
In the spayed female dog, in which blind catheterization may be difficult, the use of a sterilized otoscope speculum and light source (Figure 4-13), vaginal speculum, or anal speculum with a light source will help to visualize the urethral tubercle on the floor of the vagina. In difficult catheterizations it may be helpful to place the animal in dorsal recumbency (Figure 4-14 and 4-15). Insertion of a speculum into the vagina almost always permits visualization of the urethral papilla and facilitates passage of the catheter. Take care to avoid attempts to pass the catheter into the fossa of the clitoris because this is a blind, possibly contaminated cul-de-sac.
Catheterization of the Male Cat

Patient Preparation

Before attempting urinary bladder catheterization of the male cat, administer a short-term anesthetic (e.g., ketamine, 25 mg/kg IM), but only after a careful assessment of the cat’s physical, acid-base, and electrolyte status (see treatment of hyperkalemia in Section 1).

Figure 4-13: An otoscope speculum with attached light source provides excellent visualization of the urethral orifice in a female dog. Note the position of the otoscope handle (see Figure 4-14).

Figure 4-14: Visualization of the urethral orifice and catheterization of the urinary bladder in a female dog is accomplished using an otoscope with a sterile speculum attached. Note: The patient is in dorsal recumbency with the otoscope handle positioned upward.
In some cases, drugs to treat hyperkalemia may be required before anesthetic induction. Once the patient’s electrolyte status has been evaluated and hyperkalemia, if present, addressed appropriately, anesthesia can be induced with a combination of propofol (4 to 7 mg/kg intravenously [IV]) and diazepam (0.1 mg/kg IV); then the patient is intubated and maintained on gas anesthesia.

**Technique**

Place the anesthetized patient in dorsal recumbency. Gently grasp the ventral aspect of the prepuce and move it caudally in such a manner that the penis is extruded. Withdraw the penis from the sheath and gently pull the penis backward. Keeping sterile catheters in a freezer will help them become more rigid to facilitate passage into the urethra. Pass a sterile, flexible plastic or polyethylene (PE 60 to 90) catheter or 3- to 5-inch, 3.5F urethral catheter into the urethral orifice and gently into the bladder, keeping the catheter parallel to the vertebral column of the cat.

*Caution: Never force the catheter through the urethra.* The presence of debris within the urethral lumen may require the injection of 3 to 5 mL of sterile saline to back-flush urinary “sand” or concretions so that the catheter can be passed. In some instances the presence of cystic and urethral calculi will prevent the passage of a catheter into the urethra. For this reason a lateral radiograph of the penis, with the patient’s hindlimbs pulled caudally, may help document the presence of a urethral stone.

**Catheterization of the Female Cat**

**Patient Preparation**

Urinary bladder catheterization of the female cat is not a simple procedure. When indicated, and after a preanesthetic examination has been performed, attempt the technique only in the anesthetized cat. Urinary bladder catheterization can be accomplished with the use of a rubber or plastic, side-hole (blunt-ended) urinary catheter. The same catheter type used in male cats is effective in female cats. Instilling lidocaine 0.5% has been recommended as a means of decreasing sensitivity to catheter insertion in sedated (not recommended) cats. Cleanse the vulva with an appropriate antiseptic.
Technique
Catheterization can be accomplished with the cat in dorsal or ventral recumbency. Experience and size of the cat dictate which technique works best.

After cleansing of the perineum and vaginal vault, place the patient in sternal recumbency, and gently pass the catheter along the ventral floor of the vaginal vault. Conversely, if the patient is placed in dorsal recumbency, direct the catheter dorsally along the ventral vaginal floor. If a catheter cannot be placed blindly, a small otoscopic speculum can be placed into the vagina, and the catheter pushed into the urethral papilla once it is visualized directly.

Indwelling Urethral Catheter

Patient Preparation
For continuous urine drainage in the awake, ambulatory patient, use a closed collection system to help prevent urinary tract infection. A soft urethral or Foley catheter can be used, and polyvinyl chloride tubing should be connected to the catheter and to the collection bag outside the cage. The collection bag should be below the level of the animal's urinary bladder. Place an Elizabethan collar on the animal to discourage chewing on the catheter and associated tubing.

Technique
The urinary bladder is catheterized as described previously. Despite the quality of care of the catheter, urinary tract infection still may develop in any patient fitted with an indwelling urinary catheter. Ideally, remove the catheter as soon as it is no longer necessary, or if there are clinical signs of a urinary tract infection or previously undiagnosed fever. A urinary catheter is generally changed after it has been in place for more than 48 hours.

Special Considerations
Observe the patient for development of fever, discomfort, pyuria, or other evidence of urinary tract infection. If infection is suspected, remove the catheter and submit urine for culture and sensitivity or determination of minimum inhibitory concentration (MIC). Previously, culture of the catheter tip was recommended to diagnose a catheter-induced infection. However, culture of the catheter tip is no longer recommended, as it may not accurately reflect the type of microorganisms in a urinary tract infection. The empiric use of antibiotics to help prevent catheter-induced infection is not recommended, as their use can allow colonization of resistant nosocomial bacteria in the patient's urinary tract.

Cystocentesis

Patient Preparation
Cystocentesis is a common clinical technique used to obtain a sample of urine directly from the urinary bladder of dogs and cats when collecting a voided, or free-catch, aliquot is not preferred. The procedure is indicated when necessary to obtain bladder urine for culture purposes. Urine that is collected by free catch has passed through the urethra and may be contaminated with bacteria, thereby making interpretation of the culture results difficult. Cystocentesis also is performed as a convenience when it is desirable to obtain a small sample of urine but the patient is not ready or cooperative.

Cystocentesis involves insertion of a needle, with a 6- or 12-mL syringe attached, through the abdominal wall and bladder wall to obtain urine samples for urinalysis or bacterial culture. The technique prevents contamination of urine by urethra, genital tract, or skin and reduces the risk of obtaining a contaminated sample. Cystocentesis also may be needed to decompress a severely overdistended bladder temporarily in an animal with urethral obstruction. In these cases, cystocentesis should be performed only if urethral
catheterization is impossible. Warning: Penetration of a distended (obstructed) urinary bladder with a needle could result in rupture of the bladder.

**Technique**

To perform cystocentesis, palpate the ventral abdomen just cranial to the junction of the bladder with the urethra, and trap the urinary bladder between the fingers and the palm of the hand. Use one hand to hold the bladder steady within the peritoneal cavity while the other guides the needle. Next, insert the needle through the ventral abdominal wall into the bladder at a 45-degree angle (Figure 4-16). Although this procedure is relatively safe, the bladder must have a reasonable volume of urine, and the procedure should not be performed without first identifying and immobilizing the bladder. For the procedure to be performed safely and quickly, the patient must be cooperative. If collection of a urine sample by cystocentesis is absolutely necessary, sedation may be indicated to restrain the patient adequately for the procedure.

**Special Considerations**

Generally, cystocentesis is a safe procedure, assuming the patient is cooperative and the bladder can be identified and stabilized throughout the procedure. However, injury and adverse reactions can occur. In addition to laceration of the bladder with the inserted needle (patient moves abruptly), the needle can be passed completely through the bladder and into the colon, causing bacterial contamination of the bladder or peritoneal cavity. There is also risk of penetrating a major abdominal blood vessel, resulting in significant hemorrhage.

**Additional Reading**

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Crow S, Walshaw S: *Manual of clinical procedures in the dog, cat, and rabbit*, ed 2, Philadelphia, 1997, Lippincott-Raven.

Kruger JM, Osborne CA, Ulrich LK: Cystocentesis: Diagnostic and therapeutic considerations, *Vet Clin North Am Small Anim Pract* 26(2):353–361, 1996.
SKIN BIOPSY

Patient Preparation

Obtaining a skin biopsy from abnormal skin only to receive a nondiagnostic result as reported from a pathologist suggests that improved biopsy technique may result in collecting a specimen with higher diagnostic value. The following guidelines apply when skin biopsies are performed:

- Consider obtaining multiple samples from multiple sites, which is especially useful when different stages of similar lesions are identifiable.
- Do not perform a surgical scrub before collecting the sample; shaving the hair away is fine, but surgically prepared skin removes superficial lesions that, had they been left in place, might have been diagnostic.
- Biopsy of lesions that are depigmenting should be done before they have turned white; the absence of color usually denotes absence of active skin lesions. Biopsies from completely depigmented skin are less likely to demonstrate active lesions.
- Biopsy of lesions associated with alopecia should be done in the center of the most alopecic area.
- Also, biopsy of lesions associated with alopecia should be done at junctional (between normal- and abnormal-appearing) skin.
- Consider submitting biopsy samples from completely unaffected, normal-appearing skin.
- Avoid biopsies of ulcerated skin areas.

Technique

Biopsy samples may be obtained with a scalpel blade (incisional or excisional) or via a dermatologic punch biopsy. Punch biopsy instruments are circular blades available in 4-mm, 6-mm, and 8-mm diameter sizes (Figure 4-17). Hold the punch perpendicular to the skin site of interest. A back-and-forth motion that rotates the circular blade cuts through the skin. When the skin no longer moves as the punch is rotated, the biopsy is complete and the skin sample may be removed (from the skin or from the biopsy instrument). Avoid grasping the dermis or epidermis of the sample with any instrument to prevent crushing of the sample and causing artifact. If the sample must be lifted, use the attached subcutaneous fat only.

Figure 4-17: Disposable skin biopsy punches: 4-, 6-, and 8-mm sizes are available.
Special Considerations
If the lesion of interest is deep, the punch biopsy technique may not be effective. In this situation, an incisional or excisional biopsy using a sterile No. 10 or No. 15 surgical blade is indicated. Biopsies of ulcerated skin and solitary nodules are best done by removing a wedge of skin (incisional biopsy). In some cases it is possible surgically to remove all visible, palpable parts of the lesion (excisional biopsy). Place each sample of skin in buffered formalin, using a volume that is at least 10 times that of the sample size. If particularly large areas of skin are harvested during biopsy, cut these into 1-cm thick pieces before placing into formalin. (Note: Placing larger tissue samples into formalin may result in inadequate or incomplete fixation of the tissue and the inability to properly prepare the tissue for examination).

Alternatively, it is possible, and in many cases important, to evaluate a biopsy specimen of skin or subcutaneous tissue at the time of collection. When the lesion of interest is suspected to be neoplastic, quickly differentiating between inflammatory cells and neoplasia may be possible by simply performing an exfoliative cytologic examination (described in this section) on one of the biopsy samples in addition to fixing a separate sample in formalin and submitting that for histopathologic examination.

Small biopsy samples that have been subjected to the additional handling required to make impressions on a glass slide are not good candidates for subsequent fixation and histopathologic examination. It is generally recommended to perform exfoliative cytologic and histopathologic examinations on separate samples.

Skin Scraping
Superficial Skin Scraping
Among the most common diagnostic procedures carried out on the skin of dogs and cats is a routine skin scraping. Yet despite the frequency with which this test is used, doing a skin scraping in such a manner that the sample recovered maximizes the opportunity to establish a diagnosis can be anything but routine. A skin scraping, properly done, does require using consistent techniques appropriate to the suspected diagnoses, and as such, superficial or deep scrapings, or both, may be indicated. Skin scraping is indicated whenever ectoparasite infestation is suspected. Superficial scrapings are appropriate for detecting mites that live on the skin surface, such as *Cheyletiella* species and *Otodectes cynotis*, as well as mites that burrow within the outermost layers of skin (stratum corneum), such as *Sarcoptes* species and *Notoedres cati*.

Patient Preparation
Because the area to be scraped is relatively large (≥2 cm²), shave dogs and cats with long hair coats before attempting the procedure, unless *Cheyletiella* infestation is suspected.

Technique
Make the scraping over healthy-appearing skin. Do not cleanse the skin of superficial scale or crusts. The technique for superficial skin scraping entails the use of mineral oil or pyrethrin ear drops applied to a clean scalpel blade and directly onto the area of skin to be scraped. Scraping begins as a gentle motion made in the direction of the hair coat. Gradually increase the pressure of the blade against the skin with repetitive scrapings over the same area.

Special Considerations
Take care not to lacerate the skin, although minor capillary bleeding at the site is common. Transpose material collected on the edge of the blade to a clean glass slide, cover it with a coverslip, and thoroughly examine the material under low magnification for evidence of ectoparasites. Note that for mites such as *Cheyletiella* or scabies, finding just one mite or one egg is diagnostic and justifies implementing treatment.
Deep Skin Scraping

Patient Preparation
Same as described for superficial skin scraping (earlier).

Technique
A slightly different technique is indicated in dogs and cats suspected of having an infestation that includes *Demodex canis* mites. The mites are known to live predominantly in sebaceous glands and hair follicles. They can survive in the skin of animals without manifesting lesions. Hair loss and skin lesions develop where overgrowth of the mite population occurs. *Demodex* infestations can be localized or generalized; infestations can occur in either dogs or cats but the most severe, generalized infestations are much more likely to occur in young dogs.

Although both superficial and deep skin scrapings may reveal the presence of mites on the skin, deep scrapings may reveal *Demodex* mites in some patients when superficial scrapings are negative. The technique for deep skin scraping targets a small area of skin (<2 cm²). It may be helpful to apply gentle pressure to the skin or actually to squeeze the area of interest between the thumb and a finger in an attempt to force mites from the deeper to the more superficial skin. In some breeds (e.g., Old English Sheepdogs and Shar-Peis) recovering mites on a skin scraping can be particularly difficult. In such cases, when *Demodex* infestation is highly suspected but the results of repeated skin scrapings are negative, a skin biopsy is appropriate.

Alternatively, a procedure called a *trichogram* that involves pulling (plucking) a few hairs from the hair follicles using a hemostat may be diagnostic. Once the hairs have been plucked from the skin, place them on a glass slide that has been preprepared with a drop of mineral oil, add a coverslip, and examine the hair shaft under low magnification. Half of all dogs with *Demodex* infestation will have a positive trichogram.

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**EAR CLEANING: EXTERNAL EAR CANAL**

Not all dogs and cats with otitis externa require comprehensive ear flushing and debridement before or as part of otic therapy. In many cases, home treatment is sufficient to resolve the problems effectively, assuming the underlying diagnosis has been established. However, in patients with chronic or particularly severe infections, topical treatment administered by the owners at home may not be sufficient. In such cases, the external ear canal requires a careful and comprehensive cleaning before administration of topical medications.

Patient Preparation
Properly performed, flushing and cleaning of the external ear canals is not a quick procedure. Anesthetize the patient. Attempting to perform a thorough ear cleansing under sedation usually will not be successful. Once the animal is anesthetized, perform a careful otoscopic (or video otoscopic) examination to establish the integrity of the ear canal, evaluating, for example, for the presence or absence of tumors or parasites. In severe cases the tympanic membrane (ear drum) may not even be visible.

Technique
With the patient in lateral recumbency, flush the ear canal (Figure 4-18) or lavage it with warm saline initially, and then aspirate the material from the canal. If this procedure is not successful in removing the debris attached to the epithelium of the ear canal, use
ceruminolytic ear solutions to facilitate breakdown and removal of this material. A 5-minute instillation and soak is recommended, followed by thorough flushing to remove debris and the ceruminolytic material. Remove hair growing inside the ear canal with forceps. A suction apparatus is recommended for removal of debris and liquid remaining.

Reintroduce an otoscope to examine the integrity of the skin in the ear canal and to look for any evidence of stenosis, foreign body, or tumor. The flushing process is not complete until it is possible to visualize the tympanic membrane. Carefully remove any remaining debris with an otologic loop (Figure 4-19), not a cotton-tipped swab.

Repeat the procedure on the opposite ear as indicated. At the conclusion of the examination, apply appropriate topical medication into the ear canal before allowing the patient to recover from anesthesia. Systemic therapy or surgical intervention may be required in some patients for complete resolution of the problem. However, a thorough examination and cleaning is critical before actually making decisions regarding medical versus surgical intervention.

Special Considerations
Use of a cotton-tipped swab to remove debris from the external ear canal, although commonly done, is not generally recommended. Repeated attempts may ultimately force debris deeper into the external ear canal. General anesthesia and otic lavage or flushing may be required to effectively correct the problem created by the use of cotton-tipped swabs.

Additional Reading
Gortel K: Ear flushing. In Ettinger SJ, Feldman EC, editors: Textbook of veterinary internal medicine, ed 6, St Louis, 2005, Elsevier.

ENDOTRACHEAL INTUBATION
In selecting an appropriate endotracheal tube, consider the size of the animal and select a tube that has the largest diameter that can be inserted without force (Table 4-7). The length of tube selected must not extend beyond the bifurcation of the trachea (carina).
Patient Preparation

Always check the cuff of the endotracheal tube to ensure there are no leaks and that the cuff is working properly before intubation. Lubricate the selected endotracheal tube with sterile lubricating jelly before inducing anesthesia. After induction, place the patient in sternal recumbency and elevate the head.

Technique

The individual inserting the tube should grasp and extend the tongue with a piece of gauze. The tongue is extended to facilitate visualization of the larynx. Avoid excessive downward pressure on the tongue in order to prevent inadvertent laceration or injury from the lower incisors. If a laryngoscope is used, place the tip of scope at the base of the tongue. Gently press the tip of the laryngoscope ventrally to move the epiglottis and expose the glottis. Directly visualize the arytenoid cartilages, and then pass the tube through the arytenoid cartilages into the trachea using a slight twisting motion. If the arytenoid cartilage closes on
contact during an attempt to intubate, one or two drops of 2% lidocaine can be applied to
the arytenoid cartilages. Once the tube has been inserted into the trachea, never advance it
further than the carina. Doing so may result in intubation of either the right or the left main
bronchus (endobronchial intubation). Once the tube is in place, secure it in place with a
loop of ½-inch white tape or muzzle gauze.

Special Considerations

Overinflation of the endotracheal tube cuff can cause tracheal ulceration, tracheitis, hemor-
rhage, tracheomalacia, fibrosis, stenosis, and subcutaneous emphysema.

Additional Reading

Muir W, Hubbell J, Skarda R, et al: *Handbook of veterinary anesthesia*, ed 3, St Louis, 2000,
Mosby.

### ADvanced Procedures

**Abdominocentesis**

*Patient Preparation*

Abdominal paracentesis refers to the aspiration of fluid from the abdominal cavity for both
diagnostic and therapeutic purposes. Always weigh the animal before and after removing
abdominal fluid. Any subsequent gain in weight indicates a reaccumulation of abdominal
fluid. Place the animal in left lateral recumbency and restrain it in this position. Clip and
surgically prepare a 1- to 3-inch square between the bladder and the umbilicus just lateral
to the midline. If the bladder is distended, empty it before performing paracentesis. Infiltrate
the paracentesis site with lidocaine 0.5% using a 22- to 25-gauge needle. In most cases, local
anesthesia is not necessary. Abdominal puncture can be made with an 18- to 20-gauge nee-
dle (Figure 4-20).

*Technique*

Gently insert the needle through the skin and external abdominal oblique muscles while
simultaneously pushing and twisting, to push viscera away from the tip of the needle. Blind
abdominocentesis without the use of ultrasound to guide needle placement into a fluid pocket
can have negative results if there is less than 5 mL of fluid per kilogram within the peritoneal
cavity. When the abdominal puncture has been made, allow the animal to rest quietly to

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**Table 4-7**

| Body Weight (kg) | Magill Size | French Size | Internal Diameter (mm) |
|-----------------|-------------|-------------|------------------------|
| Dogs            |             |             |                        |
| 2               | 2           | 22          | 6                      |
| 4               | 4-5         | 26-28       | 8                      |
| 6               | 6-7         | 28-30       | 9                      |
| 9               | 8           | 32          | 10                     |
| 12              | 9-10        | 34-36       | 11-12                  |
| 14              | 9-10        | 34-36       | 11-12                  |
| 16              | 10-11       | 36-38       | 11-12                  |
| 18-20           | 11-12       | 38-44       | 12                     |
| Cats            |             |             |                        |
| 1               | 00          | 13          | 4                      |
| 2               | 0           | 16          | 5                      |
| 4               | 1           | 20          | 5                      |
facilitate drainage of the fluid. Some clinicians recommend tapping while the patient is in a standing position in the hope of obtaining more complete drainage. Changing the patient's position after the tapping may result in needle-tip laceration of intraabdominal organs. Aspiration may be easier if a specially adapted needle with multiple holes drilled in the shaft is used because it is less likely to become plugged with omentum. Ideally, tap four quadrants of the abdomen. If four-quadrant abdominocentesis has been performed and no fluid has been obtained, and if there is suspicion for peritonitis, diagnostic peritoneal lavage can be performed. The abdomen is clipped and aseptically scrubbed as previously described. Next, insert an over-the-needle catheter or hypodermic needle just caudal to the umbilicus, and instill 10 mL of warmed 0.9% sterile saline or lactated Ringer's per kilogram. After instillation of the fluid, walk the patient around or gently roll the patient from side to side to distribute the fluid throughout the abdominal cavity. Next, perform four-quadrant abdominocentesis as previously described. Only a small amount of the fluid infused will be obtained. The fluid will be diluted by the saline or lactated Ringer's infused, so biochemical analysis will not be accurate. However, the fluid can be examined microscopically for the presence of plant material, bacteria, WBCs, or bile pigment to help diagnose various forms of peritonitis.

Additional Reading
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Meyer DJ, Harvey J: Veterinary laboratory medicine, ed 3, St Louis, 2004, Elsevier.
Rudloff E: Abdominocentesis and diagnostic peritoneal lavage. In Ettinger SJ, Feldman EC, editors: Textbook of veterinary internal medicine, ed 6, St Louis, 2005, Elsevier.

BIOPSY TECHNIQUES: ADVANCED
Numerous biopsy techniques are available, and the selection of the appropriate technique is based on the tissue to be examined, the condition of the patient, and the skill of the examiner.
Excisional biopsy refers to the surgical removal of the entire lesion or organ with subsequent histologic examination. Excisional biopsy is used most frequently for skin lesions and cases in which an entire organ may have to be removed (such as an eye or an internal organ that has developed a tumor). Incisional biopsy refers to the surgical removal of a portion of a lesion with subsequent histologic examination. Choose a representative area of the lesion for biopsy. Include lesion margins, if possible. Needle aspiration refers to the use of needle and syringe to remove representative cells from the tissue or organ of interest. Specialized needles are available that allow removal of very small biopsy specimens that can be submitted for histopathologic examination. (See also Fine Needle Aspiration.)

**Needle Biopsy Techniques: General Considerations**

Needle biopsy or aspiration techniques refer to a variety of techniques used to obtain diagnostic tissue or cells from internal organs, including the lung, liver, spleen, pancreas, abdominal lymph nodes, and mass lesions within the abdomen and thorax. In contrast, fine-needle aspiration is a technique generally used to recover cytopathologic samples (cells only) from skin or subcutaneous tissues (e.g., superficial lymph nodes). The advantage of needle biopsy is related directly to how well the abnormal tissue has been characterized and how easily it can be identified during the procedure. In addition, depending on patient cooperation, most procedures can be performed safely with the patient sedated only. Short-term intravenous anesthesia and general anesthesia eliminate undesired patient movement during the biopsy procedure.

**Patient Preparation**

Potential lesions or abnormal tissues from which aspirate or biopsy samples are to be taken are located using palpation, radiographs, or ultrasound-guided imaging techniques. Shave the skin over the site of needle penetration and surgically prepare it. The type of sedation or anesthesia depends on the temperament of the animal and the site on which the biopsy will be performed.

**Technique**

Attach a 22-gauge needle without stylet to a 12-mL syringe prefilled with 0.5 to 1.0 mL of air. Optionally, affix a flexible extension set to the needle and connect it proximally to the syringe. Needle length may vary from 1 to 3½ inches depending on the required depth of penetration and size of the patient. Guide the needle into the tissue or organ of interest. Stabilize the tip of the needle to avoid random movements through organs, especially highly vascular tissue such as liver and spleen. Once the needle has been inserted, the aspiration technique entails withdrawing the plunger of the syringe to the 7- or 8-mL level. Hold that position for 1 to 2 seconds, and then release. Repeat the procedure. Depending on the nature of the lesion, it may not be indicated to thrust the needle into the tissue at multiple and different angles.

Neutralize the pressure in the syringe, and withdraw the needle rapidly. Expel any material within the needle onto glass slides using the air in the syringe. This same procedure can be repeated with a new needle to obtain an additional three to five samples from alternative sites. This technique allows samples to be obtained without applying negative pressure to the syringe, which may damage cells.

**Special Considerations**

Ultrasound guidance for needle aspirations from abdominal tissues greatly enhance the safety of this technique, especially when obtaining samples from smaller animals. Automatic-trigger needles such as Cook or Temno biopsy needles (14 to 18 gauge) are available for use in human beings but are seldom used in veterinary medicine. The risks associated with fine-needle aspiration include rupture of an encapsulated inflammatory process, dissemination of an infectious agent, seeding of neoplastic cells in the needle tract, and hemorrhage. Larger volumes of fluid and cells can be placed directly into a vial containing EDTA to prevent clot formation. Prepare and examine direct and sedimentation specimens.
Needle biopsy of internal organs using the Tru-Cut needle is particularly useful in patients with subcutaneous (Figure 4-21) or cutaneous masses and for localized abdominal and thoracic mass lesions and diffuse liver, kidney, and splenic disease. Serious complications, usually hemorrhage or laceration of the gallbladder (during liver biopsy), can occur when the procedure is performed blindly. Therefore ultrasound-guided needle biopsy is strongly recommended whenever a percutaneous biopsy of internal organs is performed. Additional safety factors provided by ultrasound guidance include the ability to image, and avoid, large aberrant blood vessels.

Risk of complications associated with needle aspiration of the lung is considerably higher than for most abdominal procedures. Pneumothorax can occur after a single, “clean” aspiration attempt. See Respiratory Tract Procedures for a detailed description of performing fine-needle aspiration of the lung.

**Additional Reading**

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Menard M, Papegeorges M: Fine needle biopsies: how to increase diagnostic yield, *Compend Contin Educ Pract Vet* 19:738, 1997.

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**Figure 4-21:** Mechanism of action of Tru-Cut biopsy needle for typical nodular biopsy. A small skin incision is made with a No. 11 blade to allow insertion of the instrument. 

A, With the instrument closed, the outer capsule is penetrated. 

B, The outer cannula is fixed in place, and the inner cannula with specimen notch is thrust into the tumor. Tissue then fills the notch. 

C, The inner cannula now is fixed while the outer cannula is moved forward to cut off the biopsy specimen. 

D, The entire instrument is removed. 

E, The inner cannula is pushed ahead to expose tissue in the specimen notch. (From Withrow SJ, Lowes N: Biopsy techniques in small animal oncology, *J Am Anim Hosp Assoc* 14:899–902, 1981.)
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**Skin Biopsy**

Histologic examination of diseased skin can serve as a means for diagnosis of cutaneous lesions. The causative agent often is found in acute and chronic skin infections. Punch biopsy of the skin is a quick and accurate means of removing a small sample of diseased skin for histopathologic examination.

**Patient Preparation**

Select a site that is well developed but not traumatized or excoriated. The sample should include little or no normal tissue. If the lesion (pustule, vesicle) can be identified early in its development and if the biopsy sample is taken only from the lesion, one may obtain a superior specimen. It is best not to take too large a sample that contains much normal skin; by mistake, the technician might take a section that misses the lesion. Proper selection of the biopsy site is crucial to accurate diagnosis. Carefully clip the hair from the lesion. Lightly blot the skin with 70% alcohol. Avoid superficial trauma while cleaning the skin. Inject a small subcutaneous bleb of 2.0% lidocaine to deaden the area. Special equipment needed for the biopsy includes a 4-mm, 6-mm, or 8-mm biopsy punch and 10% buffered formalin solution.

**Technique**

After the area has been anesthetized with lidocaine, press and rotate the biopsy punch through the skin until the subcutaneous tissue is penetrated. Remove the biopsy specimen by "spearing" the subcutaneous fat with a fine needle. Do not grasp the specimen with a forceps. Blot the specimen gently between two paper towels. Spread the tissue out gently (like a pancake), place the specimen epidermal side up on a piece of cardboard or tongue depressor, press the specimen gently to cause adhesion, and drop the specimen into the formalin fixative. The skin defect may be closed with one or two simple interrupted sutures. If deep subcutaneous tissue or large biopsy samples are needed, a punch biopsy is inadequate. Use a small (No. 15) scalpel blade to obtain an appropriate sample. In all cases in which skin biopsies are made, take multiple samples to increase the odds that at least one will have diagnostic lesions. Specimens submitted to laboratories should be accompanied by extensive, detailed clinical information, including a differential diagnosis. Skin biopsies routinely are stained with hematoxylin-eosin; however, periodic acid–Schiff, Gomori methenamine silver, and Verhoeff stains are used for special problems.

**Liver Biopsy**

The diagnosis of liver disease is generally confirmed on the basis of the patient’s clinical signs coupled with laboratory findings, radiography, and abdominal ultrasound. The development of a more specific diagnosis and prognosis in liver disease may be aided greatly by information obtained in a liver biopsy. Percutaneous liver biopsies are of much greater value in generalized liver disease such as cirrhosis, generalized acute hepatic necrosis, or amyloidosis than in focal hepatic disease. The major indications for performing a liver biopsy are (1) to explain an abnormal liver profile, (2) to define reasons for abnormal liver size, (3) to identify a possible liver tumor, (4) to arrive at a prognosis and rational approach to management, and (5) to identify the cause of ascites.

**Patient Preparation**

The procedures for obtaining liver tissue are numerous; however, needle biopsy of the liver, when performed properly, can be helpful. Careful physical and clinicopathologic examination should precede a liver biopsy. A normal coagulation profile should be documented on every patient undergoing liver biopsy. Detect and correct abnormalities in normal hemostatic mechanisms, if feasible, before needle biopsy of the liver. Liver biopsy should be performed only in the fasted patient and only after removal of ascitic fluid.
Percutaneous needle biopsies and fine-needle aspirations of the liver can be performed with local anesthesia in the sedated, and cooperative, patient. General anesthesia is a reasonable alternative whenever feasible. Biopsy sites in the liver can be selected best when needle biopsy techniques are used along with laparoscopy or ultrasound techniques. Blind percutaneous needle biopsies of the liver can be performed with relative safety if the liver is significantly enlarged and easily palpated. However, blind biopsies do carry the risk that the operator will be unable to determine the impact of penetrating the liver if only an abdominal radiograph and impression of abdominal palpation are available. In cases in which the liver is not palpable, blind biopsy carries significantly higher risk and should be performed only when no alternative exits.

A modified percutaneous liver biopsy can be performed by the following method. Place the animal in dorsal recumbency, and place a local block in the midline of the skin and abdomen at the caudoventral aspect of the left hepatic lobe. The incision into the peritoneal cavity should be large enough to accommodate the gloved index finger. Make a separate skin puncture site in the abdominal wall to accommodate the biopsy needle. Use the index finger manually to fix the left hepatic lobe (or other desired hepatic lobe) against the diaphragm or other adjacent structures, and insert the outer cannula and stylet through the abdominal wall in the isolated hepatic lobe. Remove the stylet, and rapidly insert the cutting prongs. If properly placed, the cutting prongs should not go through the entire hepatic lobe. Advance the outer cannula over the blades of the cutting prongs, thus entrapping the hepatic tissue material within the cutting prongs. Remove the biopsy needle. Using a wooden applicator stick, carefully place the biopsy specimen into fixative. Biopsy samples can be used to prepare slides for cytologic examination, and the biopsy needle may be cultured. Close the abdominal incision in the routine manner.

Another liver biopsy technique entails use of a Tru-Cut biopsy needle. Place the dog in dorsal recumbency. Clip a 5-cm area over the triangle formed by the xiphoid cartilage and left costal arch, and prepare the area as for aseptic surgery. Make a small paramedian incision large enough to accommodate a sterile otoscope head 7 mm in diameter. Use a sterilized halogen-illuminated otoscope speculum to visualize the liver. Pass a Tru-Cut biopsy needle through the otoscope cone to directly obtain a biopsy specimen of the liver.

**Nasal Biopsy**

The technique for performing diagnostic nasal biopsies is sufficiently complex (and bloody) that it is generally recommended to refer patients in need of this procedure to a specialty or referral hospital that has rhinoscopic and/or computed tomography capabilities. Blind biopsies of dogs and cats with chronic nasal disease, especially if associated with bleeding, can be associated with significant risk, including penetration of the cranium. No one likes nasal biopsy results that indicate “normal brain.”

**Renal Biopsy**

Renal biopsies can be valuable in confirming or eliminating a diagnosis of renal disease that is based on history, physical examination, and radiographic and laboratory data (Box 4-5). In addition, biopsy may be a way of arriving at a prognosis in generalized renal disease and a better means of evaluating the type of treatment to be instituted. Ultrasonographic guidance can prove valuable during renal biopsy for placing the needle into the tissue desired and avoiding complications.
Patient Preparation

Before renal biopsy, the animal should have a baseline coagulation profile that includes, at the very least, an activated coagulation time and platelet count. A buccal mucosal bleeding time may be indicated if there is any history of spontaneous bleeding in a patient with a normal platelet count. Obtain biopsies from the renal cortex. Administer fluids to patients before and after biopsy.

Many patients with generalized renal disease are critically ill and debilitated, and general anesthesia is contraindicated. In these cases, a neuroleptanalgesic agent may be used for sedation. If the animal is a good anesthetic risk and renal function will permit it, use inhalation anesthesia.

Technique

When bilateral renal disease is documented, select the left kidney for biopsy because it is more accessible than the right kidney. With the anesthetized patient in right lateral recumbency, surgically prepare the skin behind and below the junction of the costal arch at the level of the second and third lumbar vertebrae. Make a 2-inch paralumbar incision parallel to, but just behind, the costal arch. Dissect muscle and fascia until the peritoneum is visible. Carefully open the peritoneal cavity. Digitally feel for and examine the caudal pole of the left kidney. Guide the needle toward the posterior pole of the kidney with the index finger. Immobilize the kidney against the body wall and insert the Tru-Cut biopsy needle, with the biopsy notch exposed into the parenchyma of the kidney. Capture the biopsy specimen by sliding the outer sleeve of the needle over the (now embedded in the kidney) biopsy notch. Remove the needle and gently lift the biopsy sample from the needle and place it into formalin. Evaluate the site for hemorrhage. Once bleeding is controlled, a second biopsy specimen may be collected. Once bleeding from the biopsy site has stopped, the incision can be closed. In dogs, renal biopsy can be performed under ultrasound guidance using probes with channels for biopsy needle insertion.

Bone Biopsy

Evaluation of bone marrow is indicated in patients with evidence of persistently diminished cell counts of any or all cell lines (WBCs, RBCs, platelets) or evidence of morphologically abnormal cells in peripheral blood. Bone marrow aspiration and bone biopsy are extremely helpful but underused diagnostic procedures. The availability of inexpensive, high-quality biopsy needles makes these procedures safe and easy to perform (once experience is gained).

Conventional practice today is to obtain a bone marrow aspirate (cytopathologic examination) and a bone biopsy specimen from the same patient during the same procedure when changes in the peripheral blood justify this level of diagnostic testing. Bone marrow aspiration technique is described earlier in this section.

Two types of bone biopsy needles are available. The most commonly described procedure involves use of the Jamshidi biopsy needle, an 11- to 13-gauge needle that ranges in length from 5 to 10 cm (see Figure 4-11). The needle contains a stylet that extends beyond the needle tip by 3 to 4 mm. Because of the size of the Jamshidi needle, its use is limited to medium and large dogs. For bone biopsies in cats and small dogs, the Illinois bone marrow aspiration needle is preferred (see Figure 4-10), which is a 15- to 18-gauge needle available in lengths ranging from 2.5 to 5.0 cm.

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**BOX 4-5 CONTRAINDICATIONS TO RENAL BIOPSY**

- Coagulation abnormalities
- A single functional kidney
- Marked hydronephrosis
- Greatly contracted kidneys
- Acute pyelonephritis
- Large cysts
Patient Preparation

The patient usually is sedated or anesthetized for the procedure. Although some patients will tolerate this procedure when performed under local anesthesia only, the additional manipulation required to obtain a high-quality sample justifies sedation. In some cases the patient is sufficiently obtunded that sedation is neither indicated nor required.

Technique

The technique for bone biopsy is the same regardless of the needle used. Once the site has been selected (usually the same sites selected for bone marrow aspiration: head of the humerus, wing of the ilium, ischial tuberosity, proximal femur), clip the hair and surgically prepare the skin. Make a small stab incision in the skin over the site selected. Pass the needle, with stylet in place, through the incision and subcutaneous tissues until the needle tip makes firm contact with bone. Advance the needle using steady, increasing pressure and stable rotation. Rotation, in this case, means rotating the needle back and forth to the left 180 degrees and then to the right 180 degrees. Once the needle is situated in the bone (about 0.5 cm penetration only), stop. Carefully remove the stylet. Continue the penetration by gradually applying additional pressure and simultaneously rotating the needle.

The usual depth of penetration varies from 1 inch to as much as 3 inches. On reaching the desired depth, remove the needle by continuing to rotate as described but gradually withdrawing the needle from the bone. An obturator is provided to push the sample out of the bone. Place the core of bone directly into buffered formalin and submit it for histopathologic examination (decalcification will be required, which takes a little longer).

Special Considerations

Some authors recommend carefully rolling the bone core across a glass slide (for cytopathologic examination) before placing the bone in formalin. Most pathologists do not recommend this because additional handling of the biopsy sample can sufficiently disrupt the architecture of the tissue and compromise the quality of the biopsy (besides upsetting the pathologist). Note also that the needle can, with a little gentle manipulation, be reinserted into the hole from which the biopsy sample was obtained. Because the Illinois needle and the Jamshidi needle accommodate a syringe, it is possible to obtain (if done quickly, to prevent clotting) a bone marrow aspirate from the same site. Place that sample directly onto glass slides or (recommended) into 4% EDTA and mix it before making slides.

There are no specific requirements for postbiopsy care of the patient. Clean the blood from the skin using hydrogen peroxide; sutures generally are not required.

Prostate Biopsy

See Urinary Tract Procedures.

Additional Reading

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Burkhard MJ, Meyer DJ: Invasive cytology of internal organs: cytology of the thorax and abdomen, Vet Clin North Am Small Anim Pract 6:103, 1996.

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Kerwin S: Hepatic aspiration and biopsy techniques, Vet Clin North Am Small Anim Pract 25:275, 1995.

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**BLOOD GAS: ARTERIAL**

The femoral and dorsal pedal arteries can be punctured to obtain an arterial blood sample for blood gas and electrolyte analyses (see Section 1) for information on indications and interpretation of results).

**Patient Preparation**
To obtain a sample of arterial blood gas, place the patient in lateral recumbency and restrain the patient in a manner similar to that for a medial saphenous venipuncture.

**Technique**
A 25-gauge needle affixed to a tuberculin syringe is preferred for arterial puncture. Prepare the tuberculin syringe by coating it with heparin and forcing all the heparin out except for that left in the hub of the needle. Pull back on the plunger of the syringe slightly to facilitate visualizing the point at which the artery is entered. Arterial blood initially will enter the syringe without the plunger being drawn back.

For collection of blood from the femoral artery, and once the patient is sufficiently restrained, the individual collecting the arterial blood sample should palpate the medial aspect of the limb over the proximal medial femur until the femoral pulse is palpated. Direct the needle at a 30- to 45-degree angle, inserting the needle slowly, watching for a flash of blood in the hub of the needle (Figure 4-2). Gradually withdraw the plunger to facilitate blood entering the syringe. Collect 0.4 to 0.5 mL and immediately submit the blood for analysis, or place it on ice until the analysis can be performed.

To obtain blood from a dorsal pedal artery, place the patient in lateral recumbency and extend the rear limb as for a medial saphenous blood sample collection. The person obtaining the blood sample should pull the paw of the down leg in the nondominant hand toward his or her body, rotating the limb slightly in a medial direction to palpate the arterial pulse. Palpate the pulse in the dorsal pedal artery on the dorsomedial aspect of the tarsus. Gently insert the needle at a 30-degree angle into the artery, watching carefully for a flash of blood into the syringe. When the necessary amount of blood has filled the syringe, remove the needle and place pressure over the site of arterial puncture for a minimum of 2 minutes.

Evacuate excess air from the syringe and needle, and cap the needle with a red rubber stopper to prevent air from entering the needle and syringe. Place the sample on ice until analysis, if arterial blood gas analyses cannot be performed immediately.

**Figure 4-22:** Technique for collecting arterial blood from the dorsal pedal artery of a dog.
**Surgical Cutdown**

In the event that percutaneous access to a peripheral artery is not possible, the femoral artery can be isolated and prepared for surgical cutdown. After appropriate aseptic skin preparation, make a 4- to 5-cm incision in the skin over the femoral artery. Find the caudal edge of the sartorius muscle by blunt dissection and then reflect it anteriorly to expose the underlying femoral artery, vein, and nerve. Taking care to avoid tearing any vessel branches, gently isolate up to 2 cm of the femoral artery from the surrounding fascia. Visually direct the needle into the artery at this point. Alternatively, catheterize the artery in the event repeated arterial samples are required. Elevate the femoral artery by preplacing two stay sutures beneath the artery and then elevating the vessel to the level of the skin. Insert a long catheter-over-the-needle system into the lumen of the artery without penetrating the deep wall. Gently insert the catheter into the vessel, remove the needle, and cap and flush the catheter. Close the incision and affix the catheter to the skin via a tape tag sutured to the skin.

**Additional Reading**

Crow S, Walshaw S: *Manual of clinical procedures in the dog, cat, and rabbit*, ed 2, Philadelphia, 1997, Lippincott-Raven.

Davis H: Venous and arterial puncture. In Ettinger SJ, Feldman EC, editors: *Textbook of veterinary internal medicine*, ed 6, St Louis, 2005, Elsevier.

Shiroshita Y, Tanaka R, Shibazaki A, et al: Retrospective study of clinical complications occurring after arterial punctures in dogs: 111 cases, Vet Rec 146(1):16–19, 2000.

**Cerebrospinal Fluid Collection**

The collection of CSF is an important diagnostic procedure indicated for patients suspected of having significant intracranial or certain spinal diseases. However, it is our opinion that the technique to safely perform this procedure requires hands-on training and, preferably, prior experience before attempting the procedure in a clinical patient. Attempting to perform CSF collection from a written description in a text is not recommended. Although this procedure is generally safe when performed correctly, significant injury and even death are possible, despite the experience of the individual performing the procedure.

**Electrocardiography**

The electrocardiogram provides a fast, efficient way to obtain considerable data about a patient's cardiovascular status. Electrocardiography is a clinical test and must be correlated with clinical findings. Keep in mind that an electrocardiogram measures only electrical activity of the heart as seen on the body surface at any one instant. Electrical disorders of the myocardium can be transient or intermittent and, as such, can be missed on a single electrocardiogram.

**Interpretation of the Electrocardiogram**

Read each electrocardiogram using a definite system. Begin by examining the lead II rhythm strip: Is there a P wave for every QRS complex? Is there a QRS complex for every P wave? Do all the P waves look alike? Do all the QRS complexes look alike? Are the P wave and QRS complex consistently related to each other?

**Box 4-6  Indications for Performing an Electrocardiogram**

- Detect enlargement of any of the cardiac chambers
- Diagnose cardiac arrhythmia
- Identify effects of electrolyte imbalances, especially potassium
- Monitor response to and direct cardiac drug therapy
- Develop prognoses (degree of change in heart function over time)
If the answer to any of these questions is no, proceed to identify the abnormality. Next, determine the rate, rhythm, and wave character—that is, evaluate measurements of the P wave, PR interval, and QRS complex. Evaluate the ST segment, T wave, and QT interval. Use all leads to determine the axis and any miscellaneous criteria.

**Heart Rate**

Depending on the type of electrocardiographic equipment used, there are several methods for determining heart rate from the electrocardiographic tracing. Many electrocardiographs compute the heart rate and print that on the tracing. However, in patients with a significant dysrhythmia, these calculations can be flawed and should be verified manually when a question exists. Small linear lines or demarcations at the top of the electrocardiogram paper can be used to determine the heart rate. At a paper speed of 50 mm/second, the time between adjacent marks is 1.5 seconds. Counting the number of QRS complexes (or R waves) between just two of these divisions and multiplying by 20 equals the heart rate in beats per minute (Figure 4-23). For those inclined to higher mathematics, the heart rate also may be determined by counting the number of small squares between R waves (at a paper speed of 50 mm/sec) and then dividing into 3000 (Box 4-7).

**Heart Rhythm**

The normal heart rhythm is sinus in origin. For every QRS complex there is a P wave (Figure 4-24). The P waves are related to QRS complexes (P-P interval is constant). Sinus arrhythmia, sinus arrest, and wandering pacemaker are normal rhythm variations. In sinus arrhythmia, the P-P interval is irregular. The pauses are never longer than twice the usual P-P interval (Figure 4-25). A wandering pacemaker means that the P waves vary in height and may even be negative temporarily (Figure 4-26). *Sinus arrest* is defined as a prolongation of the P-R interval longer than twice the usual P-P interval.

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**Figure 4-23:** Using the electrocardiogram to determine heart rate. The distance between R waves is 20 small boxes: 3000/20 = 150 beats/min. (Paper speed is 50 mm/sec.)

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**Box 4-7 Normal Heart Rate**

| **Dog**                    | **Cat**         |
|----------------------------|-----------------|
| Large dogs: 60 to 100 beats/min | Puppies: Up to 220 beats/min |
| Medium-sized dogs: 80 to 120 beats/min | Domestic cats: 140 to 250 beats/minute |
| Small dogs: 90 to 140 beats/min |                 |
4 DIAGNOSTIC AND THERAPEUTIC PROCEDURES

Normal Electrocardiogram Measurements

P wave
The normal P wave is 0.04 second × 0.4 mV (two boxes wide × four boxes tall) for the dog and 0.04 second × 0.2 mV for the cat. In P mitrale (left atrial enlargement), the P wave is wider than 0.04 second. In P pulmonale (right atrial enlargement), the P wave is taller than 0.4 mV for the dog and 0.2 mV for the cat.

PR Interval
The PR interval is measured from the beginning of the P wave to the beginning of the QRS complex. The normal interval is 0.06 to 0.13 second (3 to 6.5 boxes wide) for the dog and 0.06 to 0.08 second for the cat. In first-degree atrioventricular heart block, the PR interval is prolonged. The PR interval is sometimes useful in monitoring the effects of digitalis therapy.

Figure 4-24: Normal lead II QRS complexes in an adult dog.

Figure 4-25: A, Mild sinus arrhythmia in a dog. There are P waves for every QRS complex, and P waves are related to the QRS complexes, which make this a sinus rhythm. The variation of the R-R intervals also is visible. An irregular sinus rhythm is a sinus arrhythmia. B, Sinus arrhythmia in the cat. (From Edwards NF: Bolton's handbook of canine and feline electrocardiography, ed 2, Philadelphia, 1987, WB Saunders.)
The QRS complex duration is measured from the beginning of the Q wave to the end of the S wave. Normal duration is up to 0.04 second in cats, 0.05 second in small dogs, and 0.06 second in large dogs. A QRS complex that is too wide indicates left ventricular enlargement (Figure 4-27). An R wave that is too tall indicates left ventricular enlargement. The amplitude is measured from the baseline to the top of the R wave (Figure 4-28). The normal R wave can be up to 0.8 mV tall in cats, 2.5 mV in small dogs, and 3.0 mV in large dogs.

Figure 4-26: A, The wandering pacemaker in this recording is suggested by the slightly negative P waves in some of the complexes. Negative P waves of this nature result from vagal depression of the sinoatrial node and the development of a junctional atrioventricular nodal rhythm. B, Marked sinus arrhythmia and a wandering pacemaker result in a decreased heart rate (increased R-R interval) and negative P waves in the fifth complex. As the pacemaker returns to the sinoatrial node, the rate increases, and positive P waves of varying amplitude result in the sixth and seventh complexes.

QRS Complex

The QRS complex duration is measured from the beginning of the Q wave to the end of the S wave. Normal duration is up to 0.04 second in cats, 0.05 second in small dogs, and 0.06 second in large dogs. A QRS complex that is too wide indicates left ventricular enlargement (Figure 4-27). An R wave that is too tall indicates left ventricular enlargement. The amplitude is measured from the baseline to the top of the R wave (Figure 4-28). The normal R wave can be up to 0.8 mV tall in cats, 2.5 mV in small dogs, and 3.0 mV in large dogs.

Figure 4-27: In these two examples of left ventricular enlargement, the QRS complexes have normal configuration but are too wide. A, This QRS complex from a miniature poodle is 0.07 second (three boxes) wide. B, This QRS complex from a Doberman Pinscher is 0.09 second (four boxes) wide. A small dog such as the Poodle should not have a QRS complex wider than 0.05 second (wider), and the larger dog’s QRS complex should not exceed 0.06 second (three boxes). Because each dog’s QRS complex is too wide, left ventricular enlargement is diagnosed in both cases. The Doberman Pinscher has no P waves because it is in atrial fibrillation. (Paper speed is 50 mm/sec; 1 cm equals 1 mV.) (From Edwards NF: Bolton’s handbook of canine and feline electrocardiography, ed 2, Philadelphia, 1987, WB Saunders.)
ST Segment

The ST segment is between the end of the S wave and the beginning of the T wave. Normally the ST segment lies on the baseline and then dips into the T wave. Slurring of S into T indicates left ventricular enlargement and is seen when the S wave slurs into the T wave and no ST segment is discernible. The ST segment is elevated if it lies more than 0.1 mV (one box) above the baseline (>0.2 mV in CV_{LL} and CV_{LU}). Elevation of the ST segment may occur with hypercalcemia or myocardial hypoxia. The ST segment is depressed if it lies more than 0.1 mV (one box; >0.2 mV in CV_{LL} and CV_{LU}) below the baseline. Depression of ST may be seen with myocardial ischemia, hypoxia, or hypocalcemia.
QT Interval
The QT interval is measured from the beginning of the Q wave to the end of the T wave. The normal interval is 0.14 to 0.22 second (7 to 11 boxes wide) in dogs and up to 0.16 second in cats. A lengthened QT interval may be seen with hypokalemia or hypocalcemia. The QT interval varies with heart rate and tends to be prolonged when bradycardia occurs. A decreased QT interval may be seen with hypercalcemia.

Mean Electrical Axis
The mean electrical cardiac axis measures the direction (vector) of the cardiac ventricular impulse during depolarization. Therefore the QRS complex is examined in leads I, II, III, aV_{R}, aV_{L}, and aV_{F}. These six leads determine the axis. They are arranged in a manner known as Bailey’s hexaxial lead system (Figure 4-29). The procedure is as follows:
1. Find an isoelectric lead—that is, a lead for which the total number of positive (upward) and negative (downward) deflections of the QRS complex is equal to zero (Figure 4-30).
   When there is no perfectly isoelectric lead, use the one that comes closest.
2. Find the lead that is perpendicular to the isoelectric lead: lead I is perpendicular to aV_{F}; lead II is perpendicular to aV_{L}; and lead III is perpendicular to aV_{R}.
3. Determine whether the perpendicular lead is positive or negative on the patient’s electrocardiogram. If the perpendicular lead is negative, the axis is at the negative end of that lead (each lead has a plus and a minus pole marked). If the perpendicular lead is positive, the mean electrical axis is at the positive end of the perpendicular lead. For example, if aV_{L} is isoelectric (normally it is), lead II is its perpendicular. If lead II is positive on the electrocardiogram, the axis is +60 degrees. If lead II is negative on the electrocardiogram, the axis is −120 degrees.

Figure 4-29: Bailey’s hexaxial reference system. The lead axes are marked in 30-degree increments from 0 to 180 degrees and from 0 to −180 degrees. The six leads are marked with a plus sign at the positive electrode and a minus sign at the negative electrode. Note that in the leads I, II, III, and aV_{F} the polarity and the angle of the leads are positive or negative simultaneously. Leads aV_{F} and aV_{L} are positive at the positions of −150 degrees and −30 degrees, respectively, because the positive electrodes for those leads lie in the negative 0- to −180-degree zone. (From Ettinger SJ, Suter PF: Canine cardiology, Philadelphia, 1970, WB Saunders.)
Normal Mean Electrical Axis

The mean electrical axis in the normal dog is +40 to +100 degrees; for the cat it is more variable, at 0 to ±180 degrees. Right axis deviation (axis more than +100) indicates right ventricular enlargement in the dog (Figure 4-31). Left axis deviation (axis 0 to +40 degrees) indicates left ventricular enlargement in the dog. When there is biventricular enlargement, the axis usually remains normal. Axis determinations are of less value in the cat because the normal range is so wide (Boxes 4-8 to 4-10).

Figure 4-30: In each of these three leads, the total of the positive and negative deflections equals zero. Each is considered an isoelectric lead.

Figure 4-31: The mean electrical axis in the frontal plane of this electrocardiogram recorded from a Wire Fox Terrier with pulmonic stenosis is approximately 165 degrees. (From Edwards NJ: Bolton's handbook of canine and feline electrocardiography, ed 2, Philadelphia, 1987, WB Saunders.)
ADvANCED PROCEDURES

endoscopy: indicAtions And equipment RequiRements

Inappropriate use of endoscopic equipment not only can damage expensive equipment but also can cause serious injury to the patient.

Upper Respiratory Tract: Laryngoscopy and Pharyngoscopy

Endoscopy of the upper respiratory tract is among the most important advanced diagnostic and therapeutic tools used in the evaluation of patients that have stertor (snorting), reverse sneeze, stridor (wheezing), and chronic cough. Laryngoscopy is of value in the diagnosis of upper airway obstructions such as eversion of the lateral ventricles, collapsed arytenoid cartilages, hyperplasia of the vocal cords, nodules on the vocal cords, elongated soft palate, collapsed proximal trachea, and traumatic injuries to the neck. Note also, however, that a careful visual examination of the larynx in the anesthetized patient (only) can be highly valuable even without the use of endoscopic equipment—for example, for assessment of laryngeal movement in patients with laryngeal paralysis. Suspected lesions inside the larynx may be difficult to visualize with or without endoscopic equipment. Examination of the trachea and main stem bronchi requires endoscopic evaluation to assess the integrity of the airway for conditions such as collapsed trachea, mediastinal tumors, hilar lymph node enlargement, parasitic nodules (Filaroides osleri), and foreign body aspiration. In addition, tracheobronchoscopy is a valuable technique that permits culturing and cytologic examination of material from bronchi involved in chronic respiratory disease. Upper airway obstruction that is not responsive to conservative therapy is an indication for more extensive diagnostic procedures, such as bronchoscopy.

| BOX 4-8 ELECTROCARDIOGRAPHIC CRITERIA FOR LEFT VENTRICULAR ENLARGEMENT |
|---|
| 1. Left axis deviation (dog) |
| 2. QRS complex too wide (but has normal configuration) |
| 3. R wave too tall |
| 4. S-T segment slurring |
| 5. May be associated with P mitrale |

| BOX 4-9 ELECTROCARDIOGRAPHIC CRITERIA FOR RIGHT VENTRICULAR ENLARGEMENT |
|---|
| 1. Right axis deviation (dog and cat) |
| 2. Presence of an S wave in leads I, II, and III (dog only) |
| 3. S wave deeper than 0.7 mV in lead CV,LU (V,) in the dog |
| 4. May be associated with P pulmonale |

| BOX 4-10 ELECTROCARDIOGRAPHIC CRITERIA FOR BIVENTRICULAR ENLARGEMENT |
|---|
| 1. Tall R wave |
| 2. Wide QRS complex |
| 3. ST segment slurring |
| 4. Deep Q waves in lead II (deeper than 0.3 mV for the cat, 0.5 mV for the dog) |
| 5. Normal mean electrical axis |
| 6. P mitrale or P pulmonale or both |

ENDOSCOPY: INDICATIONS AND EQUIPMENT REQUIREMENTS

Note: The discussion that follows centers around indications and capabilities of endoscopy in clinical practice. The discussion is not intended to be used as a “How-To” instruction guide on performing endoscopic procedures in dogs and cats. Today, numerous types of endoscopes and accessory materials are available for use in clinical practice. Specific hands-on training and complete familiarity with the equipment package available is essential before attempting to perform any of the procedures outlined.

Inappropriate use of endoscopic equipment not only can damage expensive equipment but also can cause serious injury to the patient.
Endoscopes of varying sizes are appropriate for use in examining the larynx and trachea. However, in cats and small dogs, examination of the trachea using equipment as small as a (human) bronchoscope may limit the examination because the endoscope nearly occludes the tracheal diameter. Additional training and/or experience is recommended for performing tracheoscopy in small patients.

One of the most important endoscopic techniques performed in dogs and cats involves examination of the nasopharynx, the upper respiratory compartment above the soft palate. Sometimes called pharyngoscopy, examination entails retroflexion of a small-diameter endoscope (e.g., bronchoscope) 170 to 180 degrees to allow visualization of the space between the posterior nares (choanae) and the larynx (Figure 4-32). This is a common location for

Figure 4-32: A, The appearance of the normal choanae (posterior nares) in a cat. B, The appearance of the choana of a cat with a posterior nasal mass diagnosed as lymphoma.
foreign body entrapment and occasional tumor development in cats and dogs (Figure 4-33). Pharyngoscopy is the only effective means of examining this portion of the upper respiratory tract in patients that have a history of stertor (snorting) and so-called "reverse sneeze."

**LOWER RESPIRATORY TRACT: BRONCHOSCOPY**

Endoscopic examination of the bronchi and lower airways is a highly diagnostic, occasionally therapeutic procedure indicated in patients presented with persistent cough. As in all endoscopic procedures, the patient is anesthetized for the examination. However, examination of the lower respiratory tract requires considerable attention to patient oxygenation and respiratory status during the examination. The requirement for oxygen to be administered throughout the procedure may be a significant limiting factor unless special accessories are used. In the ideal situation, the patient is a medium- to large-sized dog and the endoscope can be passed through the endotracheal tube using a T adaptor while oxygen and anesthetic are administered simultaneously.

However, in cats and small dogs it is usually not possible to pass an endoscope through the endotracheal tube. The procedure must be done by passing the endoscope directly into the trachea to the level of the right and left main bronchi and probably not much farther. Supplemental intravenous anesthetic is likely to be required because of the time required to complete the examination. Training and/or experience is essential before performing bronchoscopy, particularly in cats and small dogs.

The greatest advantage in performing bronchoscopy is to visualize the integrity of the trachea and, to a limited extent, the lower airways. Airway collapse, not visible on conventional radiography, can be strikingly apparent. Foreign body entrapment, tumors, respiratory parasites, and airway trauma also can be identified with bronchoscopy. In addition, the bronchoscopic examination allows for collection of cytologic samples from discrete areas (airways) within the lower respiratory tract. The ability to perform BAL in patients with reactive airway disease, subclinical or clinical infections, and certain types of tumors can be highly diagnostic.

**GASTROINTESTINAL ENDOSCOPY**

Flexible fiberoptic endoscopy is a noninvasive, atraumatic means of visualizing the mucosal surfaces of the esophagus, stomach, and colon. Flexible endoscopes are available from several companies at a wide range of prices. To minimize the risk of injury to the animal and
to reduce the possibility of damage to the endoscope, place animals undergoing endoscopic examination under general anesthesia after routine preanesthetic preparation. A fast of 12 to 24 hours is recommended for most patients undergoing upper gastrointestinal endoscopy. However, for patients with indications of delayed gastric emptying, a longer fast (24 to 48 hours) may be needed to empty the stomach completely. In preparation for colonoscopy, a 24- to 48-hour fast is recommended. Give a high warm-water enema the evening before and again 2 to 4 hours before the procedure. Give such enemas until the return is clear.

**Esophagoscopy**

The clinical signs indicating esophageal disease and a potential benefit of esophagoscopy include repeated regurgitation, excessive drooling, ballooning of the esophagus, anorexia or dysphagia, and recurrent pneumonia. Esophagoscopy allows visualization of the mucosal lining of the esophagus and makes it possible to detect inflammation, ulcerations, dilations, diverticula, strictures, foreign bodies, tumors, and parasite infestations.

**Gastroscopy and Duodenoscopy**

Endoscopic examination of the mucosal aspect of the stomach is indicated when the clinical signs or physical findings suggest the presence of gastric disease or when there is a need for confirmation or clarification of radiographic findings. In most cases, persistent vomiting is the chief complaint. Other clinical signs suggestive of serious gastric disease include hematemesis, melena, weight loss, anemia, and abdominal pain. Gastroscopy allows visualization of the mucosal lining of the stomach and enables detection of inflammation, ulceration, foreign bodies, and tumors. In most dogs and cats the endoscope can be passed into the proximal duodenum. Depending on the patient size and length of the scope, it may be possible to evaluate as much as 12 inches or more of the proximal duodenum.

**Colonoscopy**

Colonoscopy is endoscopic examination of colon, rectum, and anus. The technique is helpful in the definitive diagnosis of lower bowel lesions, such as granulomatous colitis, foreign bodies, tumors, lacerations, and other mucosal abnormalities. The primary indication for colonoscopy is the presence of signs of large bowel disease, which typically include tenesmus and the passage of small, frequent stools containing fresh blood or excess mucus. Endoscopic examination of the colon allows direct visualization of the effects of mucosal inflammation, ulceration, mucosal polyps, malignant neoplasia, and strictures. Histologic examination of mucosal biopsy material will confirm the diagnosis of colonic disease.

The large bowel must be empty for the colonic mucosa to be visualized. The bowel can be emptied by withholding food for 24 hours and performing a colonic irrigation the evening before and again 2 hours before the examination. The material used for the enema must be nonirritating and nonoily. Mildly hypertonic saline solutions such as Fleet enemas work well if given 2 hours before examination so that gas and fluid can be passed completely. However, do not use Fleet enemas in cats or small dogs.

If the general physical condition of the animal is poor and withholding food is not possible, feeding a low-residue diet for 12 to 18 hours before colonoscopy can be helpful. This diet could consist of cooked eggs, small amounts of cooked beef or chicken, and small amounts of carbohydrates, such as a slice of toast or ¼ to ½ cup of moist kibble. Maintain good hydration. If all food is contraindicated, oral electrolyte solutions such as Gatorade (PepsiCo, Purchase, New York) can be used to maintain hydration without moving solids through the intestinal tract.

Give the animal a short-acting anesthetic and place the animal on a tilted table in lateral recumbency with the hindquarters elevated. Perform a digital examination of the rectum and pelvic cavity to ensure that there are no strictures, polyps, or other obstructions. Lubricate the proctoscope thoroughly with water-soluble jelly and pass it gently through the anal sphincter. Press the proctoscope forward slowly and carefully with a spiral motion. If any resistance is encountered, stop the motion, remove the obturator, and inspect the
bowel to determine the cause of the resistance. If possible, replace the obturator and continue forward motion until the instrument is passed its full length. Withdraw the obturator, and observe the mucosa.

The major portion of the examination is conducted as the instrument is withdrawn. To view the colonic and rectal walls completely, one must move the anterior end of the proctoscope around the circumference of a small circle while withdrawing the proctoscope. Occasional insufflation with the inflating bulb is helpful in smoothing out folds of tissue. Repeated instrumentation may produce petechiae and minor hemorrhages that are not pathologic. For examination of the terminal rectum and anus, the Hirschman anoscope provides adequate, convenient visualization.

Newer techniques for visualizing the upper and lower gastrointestinal tract are being used in dogs. The flexible fiberoptic endoscope enables one to visualize and photograph the esophagus, colon, and stomach. One is able not only to visualize lesions of the gastrointestinal tract directly but also to assess motility, take biopsies of lesions, and remove foreign bodies.

**Vaginoscopy**

The ability to visualize directly the vestibule, the vagina to the level of the cervix, and the urethral orifice in female dogs is of particular value in evaluating patients with known or suspected congenital urinary tract disorders, such as incontinence or ectopic ureters and vaginal strictures (congenital or traumatic). Numerous vaginal malformations and chronic infections cause visual changes that are identified easily during endoscopic examination. Frequently the procedure can be conducted in the standing awake patient. Sedation or general anesthesia is indicated when extensive manipulation, catheterization of the bladder, or a vaginal biopsy are indicated. Position the sedated or anesthetized patient in dorsal or ventral recumbency to facilitate orientation during the procedure. If catheterization of the urinary bladder is required during the procedure, dorsal recumbency seems to facilitate visualization of the urethral papilla and insertion of the catheter.

Vaginoscopy entails use of a relatively small, flexible endoscope 4 to 6 mm in diameter or a 2- to 3-mm rigid scope. The flexible scope offers the advantage of a larger biopsy channel and the ability to view the lateral vaginal wall easily. Vaginoscopy is considered an invasive procedure and should be conducted under sterile conditions. Before insertion of the sterilized endoscope, the vulva should be free of obvious debris, should be clipped if necessary, and should be cleaned gently with a surgical soap and rinsed. Insert the scope such that initial position of the tip of the scope is directed toward the anus. As insertion proceeds, the tip of the endoscope reaches the horizontal portion of the vestibule and vagina. When feasible, pass the scope to the level of the cervix. Slight insufflation of the vagina may be useful in dilating the vagina, greatly facilitating the examination. Conducting the examination from the level of the cervix caudally is recommended. This maximizes the ability to visualize critical anatomic features.

**Cystoscopy**

The relatively recent introduction of very small (2-mm diameter) flexible and rigid endoscopes into veterinary medicine allows visual examination of the urethra, trigone, urinary bladder, and right and left ureterovesicualar junctions of female dogs and even cats. Such examinations are most useful when obstructive lesions (tumor or calculi) of the urethra or trigone are suspected. Visual examination of the interior surface of the bladder and the capability of collecting biopsy samples make this a particularly useful diagnostic tool in the hands of the experienced clinician.

**Additional Reading**

Guilford WG, Center SA, Strombeck DR, et al: Strombeck's small animal gastroenterology, ed 3, Philadelphia, 1996, WB Saunders.

Holt DE: Laryngoscopy and pharyngoscopy. In King LG, editor: Textbook of respiratory disease in dogs and cats, St Louis, 2004, Elsevier.
Numerous techniques are described for administering calories and nutrients to patients that are unable or unwilling to take in, chew, or swallow food. One method, intravenous hyperalimentation, is reserved for patients that are not able to tolerate any food being introduced via the gastrointestinal tract and represents a radical, and ideally transient, departure from normal. However, enteral feeding, which is always preferable to intravenous hyperalimentation, allows the clinician several options for administering food directly into the gastrointestinal tract. Consideration of several variables is critical when one is initiating enteral feeding programs, such as the patient’s diagnosis and attitude, the status of the gastrointestinal tract, and the ability of the patient to digest and absorb food once introduced. In addition, consideration of the type and constituency of the diet provided is important. Although the options available for enteral nutrition are much greater than those for intravenous hyperalimentation, the clinician must consider dietary requirements carefully when planning enteral nutritional support.

When evaluating enteral feeding for the individual patient, the clinician has four basic options: nasoesophageal tube, pharyngostomy tube (least recommended), esophagostomy tube, and percutaneous gastroscopy tube (which can be introduced using an endoscope or with the so-called “blind” technique). All techniques involve use of a polyurethane or silicone feeding tube. The nasoesophageal tube placement technique does not require general anesthesia, and the tube may be inserted using a topical anesthetic only. Each of the other techniques described requires that the patient be anesthetized to ensure proper and safe placement.

Nasoesophageal intubation

For temporary, short-term feeding, nasoesophageal intubation is a simple technique that works well in cats, puppies, and adult dogs. Patients that are comatose; have severe, persistent vomiting; have esophageal disease or dysfunction; or are unable to swallow are not candidates for this procedure. The objective of the procedure is to place a small-diameter tube (8F to 10F for dogs weighing more than 15 kg and 5F to 8F for small dogs and cats) through the nasal cavity into the distal esophagus. The tube does not have to enter the stomach. When measuring the tube length, measure from the tip of the nose to the eighth or ninth rib (Figure 4–34).

Administer 3 to 5 drops of a topical ophthalmic solution (0.5% proparacaine) directly into one nostril. Hold the head gently upward for a few seconds to allow the solution to reach the back of the nasal cavity. In most patients, it is desirable to wait 1 to 2 minutes and then to repeat the instillation in the same nostril. For larger dogs, 2% lidocaine solution (0.5 to 2.0 mL) gradually instilled into the nostril is an alternative technique to achieve topical anesthesia. Lubricate the tube with a thin coat of a water-soluble lubricant, such as a 2% lidocaine lubricating gel. Pass the tube into the nasal cavity while directing the tube tip medially and ventrally into the ventral meatus. The anatomic shape of a dog’s nostril usually requires directing the tip medially but almost perpendicular to the plane of the nasal cavity to facilitate insertion. Initial resistance (pressure, not pain) usually is perceived, and the patient’s head as expected quickly retracts, leaving the operator holding the tube tip some inches away from the patient’s nose. Be persistent. Repeat the procedure, as necessary, by quickly inserting the first inch or more of the tube into the nostril. With the other hand, push the nasal philtrum up, and with a finger, push the lateral portion of the nostril medially. This will help facilitate movement of the tube into the ventromedial nasal meatus. Once started, the remainder of the technique is relatively straightforward.
As the tube reaches the caudal aspect of the nasopharynx, it should pass directly into the esophagus with little or no resistance. Affix the tube remaining outside the patient to the head or face using a “butterfly” tape, gauze, suture (Figure 4-3), or skin glue (skin glue [Superglue] generally is not recommended because this can result in loss of hair and skin pigment when the glue becomes dislodged).

Caution: The tip of the tube can be introduced inadvertently through the glottis and into the trachea. Topical anesthetic instilled into the nose can anesthetize the arytenoid cartilages, thereby blocking a cough or gag reflex. I prefer to check the tube placement with a dry, empty syringe. Attach the test syringe to the end of the feeding tube. Rather than inject air or water in an attempt to auscultate borborygmus over the abdomen, simply attempt to aspirate air from the feeding tube (Figure 4-36). If there is no resistance during aspiration and air fills the syringe, it is likely that the tube has been placed in the trachea. Completely
remove the tube and repeat the procedure. However, if repeated attempts to aspirate are met with immediate resistance and no air enters the syringe, the tube tip is positioned properly within the esophagus. If there is any question regarding placement, a lateral survey radiograph is indicated.

**ESOPHAGOSTOMY TUBE PLACEMENT**

*Patient Preparation*

Less invasive and not requiring endoscopy equipment, esophagostomy tube placement in dogs and cats is an alternative technique to use in patients that have long-term feeding needs. Use a 14F to 20F rubber, polyurethane, or silicone feeding tube placed at the level of the middle of the cervical esophagus to the level of the eighth rib. The technique does require general anesthesia or, in the hands of an experienced individual, short-term intravenous anesthesia. The technique has been described in detail in textbooks (see Marks SL; Additional Reading). To place an esophagostomy tube, first assemble the necessary supplies: large curved Rochester-Carmalt forceps, clipper and clean blades, antimicrobial scrub, gauze squares, red rubber tube, permanent marker, scalpel blade and handle, needle holder, suture scissors, and nonabsorbable suture (0 nonabsorbable, cutting needle).

*Technique*

After placing the patient under general anesthesia and intubating the patient, place the patient in right lateral recumbency and clip the lateral left side of the neck from the ramus of the mandible caudally to the thoracic inlet, and dorsally and ventrally to midline. Note that the left side of the neck is preferred because of the normal anatomic location of the esophagus. However, if there is injury, infection, or mass that prevents placement of the esophagostomy tube in the left lateral cervical region, the right lateral side of the neck can alternately be used.

Next, aseptically scrub the clipped area, and push the Rochester-Carmalt forceps through the mouth into the esophagus. Direct the curved tips of the instrument laterally, so the tips can be visualized under the skin. Use care to note where the external jugular vein lies, to avoid laceration of the jugular vein. Measure the tube from the proposed site of tube entrance to the mid thorax, then label the tube with a permanent marker.

*Figure 4-36: Technique for verifying esophageal placement of the tip of a nasoesophageal tube in a cat.*
Next, open the curved tips of the instrument, and make a stab incision through the skin, through the open tips of the instrument, into the esophagus. Push the tips of the instrument through the skin incision. Grasp the distal end of the tube with the instrument, and clamp the instrument.

Pull the tube through the skin incision and rostrally out of the front of the mouth. If the tube does not come easily, usually the hinges of the forceps are caught on tissue within the oropharynx or pieces of the endotracheal tube.

Once the distal end of the tube is through the front of the mouth, push the distal end of the tube caudally into the esophagus with a finger or the instrument. As the distal end of the tube is pushed into the esophagus, pull the proximal end of the tube, to add tension to the tube. The proximal end of the tube will flip toward the patient’s nose when the tube is situated in the esophagus. The tube can be taped in place while radiographs are taken to confirm placement.

After radiographs confirm placement in the esophagus, suture the tube in place with two sutures, one purse-string and finger-trap around the tube entrance site, and another deep suture near the atlas, with a second finger-trap.

Finally, place antimicrobial ointment over the tube entrance site, and a loose bandage around the neck. Unlike gastrostomy tubes, esophagostomy tubes can be used immediately, and removed immediately, if the patient chooses to start eating voluntarily after tube placement.

**Special Considerations**

It is important that one first observe the technique being performed by someone with experience before attempting to place an esophagostomy tube for the first time. Although post-placement complications generally are limited to local irritation or minor infection at the site of the stoma in the midcervical region, tube placement into the mediastinum or subcutaneously can occur.

**Percutaneous Gastrostomy Tube Placement**

Percutaneous gastrostomy tubes are used routinely to administer nutrients and medications orally over days or weeks to cats and dogs that cannot have nutrients administered by mouth or that will not eat (e.g., because of feline hepatic lipidosis, oropharyngeal neoplasms, maxillary or mandibular fractures, oral reconstructive surgery, esophageal masses or foreign bodies, or severe pharyngitis). The percutaneous gastrostomy tube is placed so that it extends through the skin and left cranial abdominal wall of the abdomen into the body of the stomach.

**Catheter Preparation**

Catheter preparation for percutaneous gastrostomy tube is as follows:

1. Use the French-Pezzar mushroom-tipped catheter.
2. Cut off 1.5 cm of the open (distal) end of the catheter with scissors.
3. Cut 3-mm holes on either side of the 1.5-cm piece (outer flange).
4. Cut the distal end of the catheter to form a sharp bevel point.
5. Measure the length of the tube from the mushroom tip to 2 cm below the bevel.

**Preparation of the Stomach Tube**

Stomach tube preparation for percutaneous gastrostomy tube is as follows:

1. Use a smooth-ended vinyl stomach tube.
2. Measure the length of the tube needed to reach the stomach by laying the tube along the animal’s side with the rounded end 1 to 2 cm caudal to the last rib.
3. Mark the tube with an indelible marker or adhesive tape at the tip of the muzzle and cut off the excess tube.
4. Put the tube in the freezer for 30 minutes to stiffen the tube before beginning the procedure.
Placement of the Percutaneous Gastrostomy Tube

Placement of a percutaneous gastrostomy tube is as follows:

1. Clip and surgically prepare the skin over the left abdominal wall.
2. Place the mouth speculum between the right canine teeth.
3. Place the stomach tube in the esophagus to the level of the cardia.
4. Rotate the tube counterclockwise while carefully advancing it through the cardia.
5. Turn the tube back clockwise and advance the tube until it can be visualized through the abdominal wall 1 to 2 cm caudal to the last rib (Figure 4-37).
6. Rotate the tube so that the tip lies against the stomach and abdominal wall one third of the distance between the epaxial muscles and the ventral midline.
7. Make a 2- to 3-mm skin incision directly over the lumen of the stomach tube.
8. Use a Sovereign catheter (over the needle) and puncture the abdominal and stomach walls, placing the catheter inside the lumen of the stomach tube. Remove the needle (Figure 4-38).
9. Thread a long, rigid suture through the catheter and advance it through the stomach tube until the end is observed at the mouth end of the tube (Figure 4-39).
10. Carefully remove the plastic catheter from the stomach tube opening and place a hemostat clamp at the end of the suture material.
11. Remove the stomach tube over the oral end of the stiff introduction suture line.
12. Attach the open, beveled end of the French-Pezzar catheter stomach tube to a plastic Sovereign catheter using a mattress suture (Figure 4-40).
13. Force the tip of the rubber stomach tube into the large end of the Sovereign catheter.
14. Advance the catheter tube through the mouth and esophagus into the stomach by placing traction on the abdominal end of the introduction line.
15. The catheter will emerge through the skin incision, followed by the rubber tube. Grasp the tube with forceps and pull it through the incision opening (Figure 4-41, A).
16. Remove the catheter by cutting it off 2 cm below the beveled tip. Pull the rubber tube through the abdominal wall until slight resistance is felt (Figure 4-41, B).
17. Slide the outer flange over the end of the tube down to the skin level (Figure 4-42).
18. Apply antimicrobial ointment and a sterile gauze sponge over the skin incision.
19. Bandage the gastrostomy tube in place (Figure 4-43).

Figure 4-37: Locating the end of the rigid stomach tube at the left lateral abdominal wall. (From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat, and rabbit, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Rights were not granted to include this figure in electronic media. Please refer to the printed publication.
Figure 4-38: Placement of the Sovereign catheter through the abdominal and stomach walls and into the lumen of the stomach tube. (From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat, and rabbit, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Figure 4-39: Threading the introduction line retrograde through the Sovereign catheter and stomach tube. (From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat, and rabbit, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Rights were not granted to include this figure in electronic media. Please refer to the printed publication.
Figure 4-40: Suturing the introduction line to the beveled end of the gastrostomy catheter. (From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat, and rabbit, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Rights were not granted to include this figure in electronic media. Please refer to the printed publication.

Figure 4-41: Catheter-tube assembly being pulled through the mouth and esophagus and the stomach and abdominal walls. (From Crow S, Walshaw S: Manual of clinical procedures in the dog, cat, and rabbit, ed 2, Philadelphia, 1997, Lippincott-Raven.)

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Rights were not granted to include this figure in electronic media. Please refer to the printed publication.

Figure 4-42: Diagram showing the inner and outer flanges in place against stomach mucosa and skin, respectively. (From Crow S, Walshaw S: *Manual of clinical procedures in the dog, cat, and rabbit*, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Rights were not granted to include this figure in electronic media. Please refer to the printed publication.

Figure 4-43: Full abdominal bandage showing the plugged end of the gastrostomy tube emerging dorsally. (From Crow S, Walshaw S: *Manual of clinical procedures in the dog, cat, and rabbit*, ed 2, Philadelphia, 1997, Lippincott-Raven.)

Additional Reading
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Marks SL: Nasoesophageal, esophagostomy, and gastrostomy tube placement techniques. In Ettinger SJ, Feldman EC, editors: *Textbook of veterinary internal medicine*, ed 6, St Louis, 2005, Elsevier.
Mazzaferro EM: Esophagostomy tubes: don't underutilize them! *J Vet Emerg Care* 11(2):153–156, 2001.
OPHTHALMIC PROCEDURES

EVALUATION OF TEAR PRODUCTION

Tear production comes predominantly from the tarsal and conjunctival glands and from the accessory tarsal glands. The reflex tear secretors are the main lacrimal gland and the accessory lacrimal glands. The production of normal lacrimal secretions can be tested by using the Schirmer tear test, a standardized filter paper (Figure 4-44) that effectively measures the rate of tear production in millimeters per minute. Schirmer tear strips now are impregnated with a blue dye to facilitate visualization of the distance (in millimeters) that the tear migrates during the 1-minute test.

Patient Preparation
None required.

Technique
Each eye can be tested independently, or both eyes can be tested simultaneously in the cooperative patient. Carefully fold the notched end of the test strip before removing it from the plastic package. Insert the folded end into the lower conjunctival cul-de-sac (Figure 4-45) and begin the timing. Maintain the Schirmer test strip in position by gently holding the eyelids closed but not touching the paper. At the end of 1 minute, note the degree (distance) of wetting that occurred and record it in the medical record. The normal dog and cat should produce wetting over 10 to 25 mm in 1 minute for each eye. Amounts less than that are consistent with keratoconjunctivitis sicca. Amounts greater than 25 mm may be normal or may be consistent with excessive tear production, or epiphora.

FLUORESCIN STAINING OF THE CORnea

The cornea is composed of various layers of specialized avascular epithelium and stroma. The outer layer, the corneal epithelium, is a highly sensitive, thin layer overlying the corneal stroma, the thickest layer.

Descemet’s membrane is a distinct, thin layer of tissue beneath the stroma. The innermost layer of the cornea is the endothelium. Damage to the corneal epithelium occurs frequently in dogs and cats. Clinical presentation typically is characterized by blepharospasm of the affected eye with or without a visible ocular discharge or conjunctivitis.

Whenever superficial corneal injury is suspected, assessment of the integrity of the corneal epithelium is indicated. Fluorescein dye–impregnated test strips can be used to determine whether the epithelial barrier overlying the corneal stroma has been disrupted and thus can establish the presence or absence of a corneal ulcer (Figure 4-46).
Patient Preparation
None required.

Technique
The test is simple to accomplish. Moisten the dye-impregnated tip of the test strip with a drop of balanced saline solution (or commercial ocular irrigation solution). Gently allow the tip of the test paper to touch the cornea, or sclera, of the affected eye. (In patients with particularly painful, sensitive eyes, use a topical anesthetic to moisten the test strip or apply

Figure 4-45: Placement of a Schirmer tear test strip into the lower conjunctival cul-de-sac of a dog; the test strip is held in place for 60 seconds only.

Figure 4-46: Fluorescein sodium–impregnated test strip used to enhance visualization of a corneal ulcer.
the anesthetic directly to the cornea before testing.) Immediately rinse the eye with a sterile irrigation solution to remove the excess dye (the test strip has a lot of dye; be prepared to catch the excess fluid with 2 × 2-inch gauze).

Promptly examine the eye with a direct, focal light source. Evidence of green dye uptake in the stroma indicates that an ulcer is present. The absence of staining generally indicates that the corneal integrity is intact. One exception exists. The Descemet membrane will not take up fluorescein dye. A patient with a deep corneal ulcer that penetrates through the corneal stroma and allows herniation of the Descemet membrane (descemetocoele) will not demonstrate a positive stain. Careful visualization of the cornea, however, is likely to reveal the presence of such a serious, deep ulcer.

**Assessment of Nasolacrimal Duct Patent**

**Patient Preparation**

None required.

**Technique**

Fluorescein dye can also be used to assess patency of the nasolacrimal duct. To perform this examination, place a drop of fluorescein dye from a sterile fluorescein strip into the eye and add 1 or 2 drops of a sterile eye wash. After 2 to 5 minutes, examine the external nares with the aid of a cobalt blue filter or Wood light for the presence or absence of fluorescence. A clean, 2 × 2-inch white gauze square touched against the nasal planum also will pick up the green-colored dye if the duct is patent. If dye is present, the lacrimal excretory system is patent and functioning. If epiphora exists but the primary dye test indicates that the lacrimal excretory system is patent, hypersecretion of tear fluid may be implicated as the cause of the epiphora.

Irrigation of the nasolacrimal system is indicated if the primary dye test result is negative. In the dog the nasolacrimal puncta are located 1 to 3 mm from the medial canthus on the mucocutaneous border of the upper and lower lids. In the dog, use a 20- to 22-gauge (in the cat, a 23-gauge) nasolacrimal cannula (Figure 4-47). Topical anesthesia often is required. Fill a 2-mL syringe with saline, and attach the lacrimal cannula and pass it into the lacrimal puncta of the upper lid.

**Special Considerations**

Several points should be made about evaluating the nasolacrimal system. Brachycephalic-breed dogs and cats occasionally may have a negative primary dye test result, although no blockage in the nasolacrimal system exists. In flushing the nasolacrimal system of some animals, fluid may not appear at the nose; however, the animal may gag and exhibit swallowing movements, indicating that the fluid has entered the mouth and the system is patent.

**Figure 4-47:** Lacrimal cannulas used to flush the nasolacrimal ducts.
CONJUNCTIVAL SMEARS, SCRAPINGS, AND CULTURES

Patient Preparation
None required. This procedure is preferably performed without administration of topical anesthesia. Topical anesthetics not only are bacteriostatic but also may distort the cells and compromise the cytologic examination.

Technique
In performing conjunctival scrapings, use a platinum spatula (Kimura spatula), the tip of which has been sterilized. Gently scrape the inferior conjunctival cul-de-sac (Figure 4-48). Place the material on two glass slides. Fix one slide in acetone-free 95% methanol for 5 to 10 minutes, then stain the slide with Giemsa stain. Heat-fix the other slide, and apply Gram stain.

To culture the conjunctiva, use sterile cotton-tipped applicators, fluid thioglycolate medium, and blood agar medium. Evert the palpebral conjunctiva of the lower lid, and pass one side of a sterile cotton applicator, previously moistened with sterile broth or thioglycolate medium, over the palpebral conjunctival surface. Streak the swab onto a sterile blood agar plate, then place the plate in a tube of thioglycolate broth. No topical anesthesia is used before culturing because preservatives present in anesthetics can inhibit the growth of bacteria.

TONOMETRY
Glaucoma is an increase in intraocular pressure incompatible with normal ocular and visual functions. One method used to measure intraocular pressure is tonometry, in which the tension of the outer coat of the eye is assessed by measuring the impressibility, or applanability, of the cornea. Because the measurements based on tonometry involve calculations that have a wide base of variations, tonometry readings are always approximations.

Schiotz Tonometry
The Schiotz tonometer consists of a corneal footplate, plunger, holding bracket, recording scale, and 5.5-, 7.5-, 10.0-, and 15.0-g weights. The principle of the Schiotz tonometer is that the amount that the plunger protrudes from the footplate is related to the indentability of the cornea, which in turn is related to the intraocular pressure. However, use of applanation tonometry today has virtually replaced use of the Schiotz tonometer.
Applanation Tonometry
In applanation tonometry, a very small area of the cornea is flattened by a known force, usually a calibrated burst of air. The advantage of this technique over the indentation (Schiøtz) method is that the errors resulting from ocular rigidity and corneal curvature are greatly reduced. Special equipment is required to perform applanation tonometry (Figure 4-49).

Gonioscopy
The presence or absence of glaucoma, an increase in intraocular pressure, can be determined using applanation tonometry. On the other hand, gonioscopy permits one to visualize and examine the iridocorneal angle and potentially establish the cause of glaucoma. However, specific training and equipment are required not only to perform gonioscopy but also to interpret the result. This procedure is most appropriately performed by an ophthalmologist.

Additional Reading
Barnett KC, Crispin SM: Feline ophthalmology, Philadelphia, 1998, WB Saunders.
Barnett KC, Sansom J, Heinrich C: Canine ophthalmology, Philadelphia, 2002, WB Saunders.

Radiography: Advanced Contrast Studies
Gastrointestinal Studies
When considering a contrast study of the gastrointestinal tract, it is not unreasonable to question the value of doing the procedure. At issue is the fact that abdominal ultrasound and/or gastrointestinal endoscopy has largely replaced contrast radiography of the gastrointestinal tract and for good reason. Diagnostic modalities such as ultrasound (in the hands of an experienced individual) and endoscopy have a much greater diagnostic yield than the less sensitive contrast study. So why even try? Endoscopes are not available in every practice, and limited access to ultrasound equipment, much less someone who is qualified to use it, puts routine use of advanced diagnostic modalities out of reach for many practices. However, it must be appreciated that with regard to diagnostic value, a radiographic contrast study of the gastrointestinal tract is a far less sensitive diagnostic modality than abdominal ultrasound or endoscopy. The procedure for the gastrointestinal radiographic contrast study is outlined next.

Figure 4-49: Measurement of intraocular pressure using applanation tonometry.
Contrast agents available for gastrointestinal studies include barium suspension preparations or Micropaque (Guerbet, Villepinte, France), and water-soluble agents (Gastrografin [Bracco Diagnostics, Princeton, New Jersey], which is 60% meglumine and 10% sodium diatrizoate). Water-soluble agents are used if bowel perforation is suspected. Undiluted water-soluble agents are hypertonic and should be diluted at a ratio of one part Gastrografin to two parts water. No single procedure is appropriate for all gastrointestinal cases. The clinician must select procedures based on the clinical history and physical findings, apparent location of the lesion within the gastrointestinal tract, endoscopic findings, and results from other imaging studies, such as abdominal ultrasound.

**Contrast Esophagram**
The contrast esophagram also is called barium swallow. The decision to perform a contrast esophagram is based on physical evidence of dysphagia (difficulty or pain while attempting to swallow) and/or persistent regurgitation (reflux of swallowed food without effort).

**Patient Preparation**
The procedure necessitates that the animal fast for 12 hours before radiography. Remove all leashes from around the animal’s neck, and obtain survey radiographs of the thorax. In esophageal contrast studies, administer barium suspension contrast medium, 2 to 5 mL/kg body mass. Administration of barium as a contrast material is contraindicated if a perforation of the esophagus is suspected. When the esophagus has been coated with radiopaque material, take lateral, ventrodorsal, and right ventrodorsal oblique thoracic radiographs to visualize the esophagus.

**Technique**
Properly prepared, the barium should be relatively thick and of a pastelike consistency. Position the patient and cassette, and have the radiographic technique set. Give a tablespoonful of barium orally. Make the exposure when the animal takes its second swallow after the barium has been given.

For esophageal studies and barium swallows, sedation with acepromazine and buprenorphine (IV, IM, SQ) will produce no adverse alteration in gastrointestinal motility. For cats, ketamine 10 mg IV and midazolam 0.2 mg/kg (combined) can be administered intramuscularly (IM) with no significant effect in esophageal motility. Caution: Patients with significant swallowing disorders have a risk of aspiration if contrast material is regurgitated. Sedation can increase that risk.

In some cases of incomplete esophageal stricture, barium liquid will pass through the esophagus unobstructed, whereas food will not. Veterinarians should mix kibbled food with the barium in this case and allow the patient to eat the mixture just before the radiograph is taken.

**Special Considerations**
Ideally, contrast esophagrams are performed using fluoroscopy rather than conventional radiographs. In this manner it is possible not only to identify strictures and dilatations, if present, but also to obtain a dynamic study of the esophagus that provides valuable information pertaining to swallowing and esophageal motility and function and an opportunity to evaluate sphincter activity at the level of the cardia.

**Upper Gastrointestinal Tract (Stomach, Pylorus, and Small Intestine)**
Contrast studies of the upper gastrointestinal tract are used to facilitate diagnosis of persistent vomiting, hematemesis, unexplained and chronic diarrhea, suspected enteric foreign bodies, and suspected neoplasms and obstructions and for confirmation of displaced intestinal organs, as may be seen in diaphragmatic hernias.

That said, abdominal ultrasound has become sufficiently available to largely replaced the upper gastrointestinal series. With an experienced ultrasonographer, the diagnostic value of abdominal ultrasound far exceeds that derived from evaluating sequential radiographs of
a patient after oral administration of a contrast medium such as barium. In the event that ultrasound capability is not available, a contrast study of the upper gastrointestinal tract still can be performed. However, the clinician must appreciate that a barium contrast study of the stomach, duodenum, jejunum, and ileum has a low sensitivity as a diagnostic test. That is, negative findings are not expected to correlate well with the absence of clinical disease. A negative study does not rule out disease. Likewise, a contrast study of the upper gastrointestinal tract is not recognized for its ability to confirm a diagnosis of gastrointestinal tract disease, even when disease is present. Perhaps the greatest value in performing the upper gastrointestinal series in a dog or cat today centers on the need to identify a displacement of the stomach and/or small intestine because of an extraluminal mass lesion or congenital defect in the patient. In addition, the use of a microfine barium suspension may facilitate identification of intestinal ulcers, irregularities (e.g., intraluminal neoplasia), and radiolucent foreign bodies. However, variable-diameter, solid-phase radiopaque markers called barium-impregnated polyethylene spheres (BIPS) can be used to assess gastric emptying time, gastrointestinal transit times, and, to some extent, obstructive disorders.

**Technique**

If an upper gastrointestinal study is indicated, follow the technique described:

1. Ensure that the hair of the animal is free from dirt, paint, and foreign material. Bathe the animal if necessary.
2. Withhold food for 18 to 24 hours.
3. If the colon is filled with feces, administer a cleansing enema the evening before performing the procedure. In dogs, give a second enema 3 to 5 hours before the start of the gastrointestinal series.
4. At the start of an upper gastrointestinal series, obtain survey radiographs of the abdomen. Administer a barium sulfate (micropulverized) preparation by stomach tube, or induce the animal to swallow the fluids. Flavored, prepared barium suspensions are available, but they taste bad (personal experience). Dosage levels vary, but for barium suspensions, give approximately 10 mL/kg. As an alternative to barium, use an organic iodide liquid preparation. Administer 0.5 mL/kg by stomach tube. Obtain lateral and dorsoventral radiographs of the abdomen immediately after administration of the contrast material and at 30-minute, 1-hour, and 2-hour intervals. Water-soluble contrast material passes through the gastrointestinal tract in 30 to 90 minutes. Barium suspensions take 60 to 180 minutes to traverse the intestine. The colon usually is filled with barium 6 hours after oral administration and may contain barium for 2 to 3 days after administration.

Barium contrast radiography is contraindicated if perforation of the stomach or upper gastrointestinal tract is suspected. In these cases, use water-soluble contrast media such as the oral diatrizoates because leakage into the abdomen will produce no foreign body granuloma. In addition, do not administer barium sulfate when an obstruction of the lower bowel may be present. In these cases, barium may only contribute to the obstipation.

The following radiographic views are recommended after administration of radiographic contrast material:

1. Immediately after administration of contrast material, obtain ventrodorsal, right lateral, and left lateral views. The right lateral view shows the pylorus of the stomach filled with barium, and the left lateral view shows the cardia and fundic portion filled with barium. The objective is to evaluate the distended stomach and initial gastric emptying.
2. Twenty to 30 minutes after administration of contrast material, obtain ventrodorsal and right lateral views to assess the stomach, pyloric emptying, and the proximal duodenum.
3. Sixty minutes after administration of contrast material, repeat the ventrodorsal and right lateral recumbency views to assess the small intestine.
4. Two hours after administration of contrast material, repeat the ventrodorsal and right lateral views to evaluate passage of contrast material into the colon and complete emptying of the stomach; contrast material should be in the terminal portion of the small intestine.
Guidelines for Passage of Contrast Material through the Gastrointestinal Tract

The passage of contrast material through the normal gastrointestinal tract is variable; however, the following guidelines have been suggested:

1. Contrast material is in the duodenum within 15 minutes in most patients. Excitement can delay gastric emptying time to 20 to 25 minutes.
2. Contrast material reaches the jejunum within 30 minutes and is within the jejunum and ileum at 60 minutes.
3. Contrast material reaches the ileocecal junction in 90 to 120 minutes.
4. At 3 to 5 hours after administration, contrast material has cleared the upper gastrointestinal tract and is within the ileum and the large intestine.

In evaluation of gastrointestinal contrast studies, consider the following criteria: (1) the size of the intestinal mass, (2) the contour of the mucosal surface, (3) the thickness of the bowel wall, (4) the flexibility and motility of the bowel wall, (5) the position of the small intestine, (6) the continuity of the opaque column, and (7) the transit time.

The Barium Enema

Clinical disorders for which the barium enema is indicated in dogs include ileocolic intussusception and cecal inversion (intussusception), mechanical and functional large bowel obstruction, invasive lesions of the large bowel, a mass outside the large bowel compressing the bowel, and inflammation of the lower intestinal tract. Barium sulfate enemas are contraindicated in suspected obstruction of the colon and rupture or perforation of the colon. However, these same disorders also can be identified by ultrasonic examination or colonoscopy, either of which is the preferred diagnostic modality over a barium enema.

Patient Preparation

Twenty-four hours before radiographs, administer a liquid diet only, preferably water. During the 18 to 24 hours before the radiographs, administer a mild high colonic enema or give a saline laxative orally. Do not give any irritating enemas within 12 hours of the scheduled radiographic examination; however, administer isotonic saline solution or plain water enemas before the examination to ensure that the bowel is clear. Obtain survey radiographs of the abdomen, and examine the colon to ensure that this portion of the bowel is clear. Sedation or anesthesia may be indicated.

Technique

Barium may be infused through a catheter into the colon or allowed to flow in by gravity through an enema bag. Do not force barium into the colon under pressure. Do not elevate the enema bag more than 18 inches above the animal.

Cuffed rectal catheters (Bardex cuffed rectal catheters, 24F to 38F, and the Bardex cuffed pediatric rectal catheter, 18F [C.R. Bard, Murray Hill, New Jersey]) can be used in dogs (Figure 4-50). For very small dogs and cats, use smaller catheters. A plastic catheter adapter and a three-way stopcock are needed. Various barium sulfate preparations can be used; however, the final concentration should be 15% to 20% w/v. A commercially available barium enema kit is helpful.

Place the cuffed rectal catheter so that the inflated bulb is cranial to the anal sphincter. Place the animal in right lateral recumbency and fill the colon with contrast material at a dose of 20 to 30 mL/kg. Take the radiographs after infusion of a two-thirds dose of barium. If the colon is not filled, infuse more contrast agent. Obtain radiographs in the ventrodorsal and lateral positions, and determine whether the colon is distended adequately. Remove as much of the contrast material as possible from the colon, and repeat the radiographs.

Insertion of room air at 2 mL/kg into the colon facilitates the evaluation of the colonic surface. Deflate the cuff on the catheter, and remove the catheter from the rectum. Throughout the procedure of filling the colon with contrast material or air, take care not to overdistend the colon, which may lead to rupture.

When reviewing individual radiographs, look for the following radiographic lesions: (1) irregularity of the barium-mucosal interface; (2) spasm, stricture, or occlusion of the bowel...
lumen; (3) filling defects; (4) outpouching of the bowel wall caused by diverticulum or perforation; and (5) displacement of the bowel.

**Additional Reading**
Burk RL, Ackerman N: *Small animal radiology and ultrasonography: a diagnostic atlas and text*, ed 3, St Louis, 2003, Elsevier.
Hall EJ, German AJ: Diseases of the small intestine. In Ettinger SJ, Feldman EC, editors: *Textbook of veterinary internal medicine*, ed 6, St Louis, 2005, Elsevier.
Thrall DE: *Textbook of veterinary diagnostic radiology*, ed 4, Philadelphia, 2002, WB Saunders.
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**Excretory Urography**
Intravenous administration of organic iodinated compounds in high concentrations permits visualization in four phases of excretory urography: (1) the arteriogram, (2) the nephrogram, (3) the pyelogram, and (4) the cystogram (Box 4-11). The arterial phase demonstrates renal blood flow; the nephrogram demonstrates the accumulation of contrast agent in the renal tubules and is used to evaluate renal parenchyma; the pyelogram phase evaluates the urinary collecting system, including the ureters; and the cystogram reveals the collection of contrast agent in the urinary bladder.

Excretory urography does not result in any quantitative information about renal function and is not a substitute for renal function tests. The degree of visualization of contrast material within the renal excretory system depends on the concentration of iodine in the contrast medium, the technique of excretory urography performed, the state of hydration of the patient, renal blood flow, and the functional capacity of the kidneys.

**Patient Preparation**
An intravenous catheter is prepositioned.

| BOX 4-11  PATIENT PREPARATION FOR EXCRETORY UROGRAPHY |
|-------------------------------------------------------|
| 1. Have the patient fast for 12 to 18 hours.         |
| 2. Administer a cleansing enema or give a saline laxative orally 12 to 18 hours before radiography. |
| 3. Ensure that the animal’s hair is free of dirt and debris. |
| 4. Try to limit the animal’s fluid intake in the 12 hours preceding radiography. |
| 5. Empty the animal’s bladder immediately before taking radiographs. |
| 6. Take survey radiographs before administering contrast media. |
Technique
The contrast medium most commonly used is a diatrizoate or iothalamate compound. Administer 850 mg/kg of an iodine compound IV by syringe. Continuous IV “push” is indicated. Obtain a ventrodorsal radiograph at 10 seconds after injection, and repeat ventrodorsal and lateral radiographs 1, 3, 5, 15, 20, and 40 minutes after injection. This method is the current standard technique. If the patient’s blood urea nitrogen level is greater than or equal to 50 mg/dL or the creatinine level is greater than 4 mg/dL, double the dose of contrast material.

Lesions that can be detected by using intravenous urography are renal mass lesions; neoplasia; renal cysts; renal and ureteral traumatic lesions; pyelonephritis; hydroureter; hydronephrosis; renal agenesis; hypoplasia; pelvic and ureteral obstructions (calculi, blood clots); renal parasites; ectopic ureter; and duplication of the collecting system.

Retrograde Contrast Urethrography
Retrograde urethrography is a diagnostic tool used to localize diseases of the lower urinary tract of dogs and cats. This method can reveal conditions such as urethral neoplasms, strictures, trauma, calculi, or other anomalies.

Patient Preparation
The procedure involves the injection of an aqueous iodine contrast medium into the urethra through a ureteral or balloon-tipped catheter. The radiopaque contrast material is mixed to a threefold to fivefold dilution with sterile lubricating jelly to increase the viscosity. A dilution of 1:3 contrast medium with sterile distilled water or saline also can be used. Before retrograde contrast urethrography is performed, give the animal a cleansing enema. Sedation or anesthesia may be necessary.

Technique
Inject 5 to 10 mL of contrast medium. Near the end of the injection, while the urethra is still under pressure, obtain a lateral radiograph. If the urinary bladder is to be distended with contrast material or air, remove urine from the bladder. In the male dog, position the catheter so that the tip of the catheter is distal to the os penis. Inject lidocaine 1 to 2 mL into the urethral lumen to anesthetize the urethra adjacent to the balloon-tipped catheter.

In male cats, retrograde contrast urethrography can aid in defining the extent of urethral damage (stricture) or the presence of urethral calculi. In male cats, use a 4F balloon catheter or a 3.5F Tomcat open-ended urethral catheter. Insert the catheter 1.5 cm into the penile urethra. If the urethra is patent, 2 to 3 mL of contrast material will enable visualization of the urethra, but increased amounts of contrast material (2 to 3 mL/lb) injected into the bladder are needed for maximum distension of the preprostatic urethra. A voiding positive contrast urethrogram is necessary to visualize the distal (penile) urethra. Apply external pressure to the bladder (using a wooden spoon or other external compression device), and radiograph the distal urethra.

Special Considerations
Take extreme care with the amount of fluid placed in the bladder if the urethra is occluded by a balloon catheter. Overdistension of the bladder results in hematuria, pyuria, urinary bladder rupture, and mild to severe bladder inflammation. Palpate the bladder carefully during distension, and note the backpressure on the syringe used in filling the bladder.

Cystography
Cystography refers to contrast radiographic procedures that facilitate visualization of the lumen and/or contents of the urinary bladder and trigone (Box 4-12). Three procedures can be used to image the urinary bladder: positive contrast cystography, negative contrast cystography (also called pneumocystography), and double-contrast cystography (combination of positive and negative cystography performed in the same patient). Note: Many of the indications for performing contrast cystography are also indications for ultrasound
examination. Contrast cystography is most useful for characterizing congenital and acquired alterations in the normal anatomy and function of the ureters and lower urinary tract, such as ectopic ureter. Abdominal ultrasound, when available, remains the preferred method for imaging abnormalities within the bladder lumen (e.g., calculi and tumors) and changes within the bladder wall.

**Pneumocystography**

Pneumocystography, also called negative-contrast cystography, involves the insufflation of a soluble gas into the lumen of the urinary bladder to facilitate imaging of any material or tissue within the bladder lumen that otherwise would be obscured by the presence of urine or positive contrast material.

**Patient Preparation**

Prepare the patient as described previously.

**Technique**

Once a urinary catheter has been placed and the urethra is occluded, use a syringe and a three-way stopcock to inject 4 to 10 mL of carbon dioxide or nitrous oxide per kilogram. Palpate the bladder while filling it with gas to avoid overdistension or rupture. Inject air until there is pressure on the syringe barrel or leakage of air around the catheter. Replace any air that escapes during the procedure. Take lateral and ventrodorsal views of the abdomen.

**Caution:** Room air is the most accessible contrast material for pneumocystography and generally can be found in most practices. However, an increased risk of air emboli is associated with the placement of room air into the bladder under positive pressure, particularly in patients with hematuria.

**Special Considerations**

Pneumocystography is not an innocuous procedure; fatal venous air emboli have occurred in dogs and cats. This complication is seen most commonly in cases of severe hematuria. Ultrasound or positive contrast cystography is preferred over pneumocystography in such cases if a soluble gas is not available. If possible, use a gas that is readily soluble in blood (such as carbon dioxide or nitrous oxide) for bladder insufflation.

**Positive Contrast Cystography**

The injection of radiographic contrast material into the urinary bladder is referred to as contrast cystography or positive contrast cystography. When ultrasound examination is not available or not feasible, the clinical and radiographic findings noted in Box 4-13 justify the use of contrast radiography to image the bladder.

The same principles of preparation apply as for obtaining a pneumocystogram. Use a urethral catheter with a three-way valve or a small Foley catheter with an inflatable cuff. Organic iodides are the contrast material of choice and should be used in 5% to 10% concentrations.
Double-contrast Cystography

Double-contrast cystography also can be performed in patients for which a positive contrast study is not diagnostic, yet there is reasonable indication for an intraluminal lesion. In this case, the same urinary catheter as used for the contrast study, remove all remaining urine and contrast material. If necessary, inject 2 to 5 mL of an aqueous organic iodine contrast material into the bladder. Gently roll the patient over in an attempt to coat the bladder with contrast material. Then distend the bladder with air in the same manner as described for pneumocystography.

Some of the lesions routinely diagnosed with the aid of cystography are calculi (Table 4-8); neoplasia; cystitis, if proliferative changes are present; muscle hypertrophy; bladder diverticula; duplications; adhesions, especially uterine stump infection; persistent urachus; ruptures; and atonic bladder.

Additional Reading

Burk RL, Ackerman N: Small animal radiology and ultrasonography: a diagnostic atlas and text, ed 3, St Louis, 2003, Elsevier.

Osborne CA, Finco DR: Canine and feline nephrology and urology, Baltimore, 1995, Williams & Wilkins.

Thrall DE: Textbook of veterinary diagnostic radiology, ed 4, St Louis, 2002, Elsevier.

REPRODUCTIVE TRACT: FEMALE

Vaginal examination is indicated for collection of material from the mucosal wall for culture and exfoliative cytologic examination and for vaginoscopic examination of vaginal and cervical mucosa (Box 4-14).

| Calculus Composition       | Density                                      |
|----------------------------|----------------------------------------------|
| Calcium oxalate            | Radiopaque                                   |
| Calcium carbonate          | Radiopaque                                   |
| Triple phosphate           | Radiopaque—small calculi may be nonradiopaque|
| Cystine                    | Variable density—may have radiopaque stippling|
| Uric acid and urates       | Nonradiopaque                                |
| Xanthine                   | Nonradiopaque                                |
| Matrix concretions         | Nonradiopaque                                |

Table 4-8 Radiopacity of Cystic Calculi on Plain Abdominal Radiographs

From Park RD: Radiology of the urinary bladder and urethra. In O'Brien TR, editor: Radiographic diagnosis of abdominal disorders in the dog and cat, Philadelphia, 1978, WB Saunders.
Examination of the vagina for culture and cytologic or vaginoscopic examination occasionally can be performed in the cooperative patient without the use of sedation or anesthesia. An assistant is used to restrain the patient on an examination table. Bitches that can be restrained for other minor examinations (ears, teeth, toenails, anal sacs, and blood samples) often will tolerate vaginal examinations. Those that need further restraint may require sedation or administration of a short-acting barbiturate anesthetic.

**Patient Preparation**

Trim long perivulvar hair and cleanse the perineum with a germicidal or surgical scrub such as povidone-iodine. Water and germicidal soap usually will not control surface contamination by *Pseudomonas* and *Proteus* species, which frequently contaminate culture swabs. In dogs with long tail hair, it is appropriate to wrap the tail with gauze before the procedure to prevent bacterial contamination.

**Technique**

If vaginal culture is indicated, this procedure should be conducted first to avoid contamination induced by the general examination. Pass a sterile, warm vaginal speculum with only a thin coating of lubricating gel into the posterior vagina while an assistant spreads the vulva. Guide the speculum into the vagina by placing the speculum into the vulva just at the dorsal commissure of the vulva and applying pressure up and out against the commissure. Direct the speculum dorsally toward the rectum until meeting resistance, and then direct it horizontally into the cranial vagina. This procedure bypasses the clitoral fossa and enables visualization of the urethral opening and pelvic arch.

Take a guarded culture swab (swab covered by a protective plastic pipette) from its individual sterile bag and pass it inside the vaginal speculum to the anterior vagina or cervical area. Then expose the swab from the protective plastic tubing and rotate it against the mucosa. Retract the swab into the protective plastic tubing and carefully remove it from the vagina. The protected swab then may be placed back in its original sterile bag until it is processed for culture (30 minutes) or placed in Amies transport medium with charcoal. Amies transport medium with refrigerator packs and a Styrofoam-insulated mailing box will retain fastidious organisms for 72 to 96 hours. Process bacterial, *Mycoplasma*, and *Ureaplasma* cultures for potential infectious agents. Viral transport medium can be used for a separate sterile swab if viral agents such as the genital form of canine herpesvirus are suspected.

Immediately after the swabbing for culture, while the vaginal speculum is still in place, advance a clean or sterile swab moistened with sterile physiologic saline solution carefully into the anterior vagina to make a smear for cytologic examination. Gently scrape vaginal epithelial cells from the ceiling of the vagina at or cranial to the region of the external urethral orifice. Collect samples from the region of the clitoral fossa, which is lined by stratified squamous epithelium at all stages of the estrous cycle. Gently rub the swab on the vaginal mucosa. Remove the swab and roll it smoothly onto two or three clean glass slides.

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**Box 4-14 Equipment to Use for Examination of the Canine Vagina**

| Equipment                                |
|------------------------------------------|
| Sterile vaginal speculum (e.g., adjustable spreading, stainless steel, or disposable plastic; cylindric; glass, plastic, stainless steel, or nylon) |
| Sterile otoscope heads of variable size for small dogs |
| Sterile protected culture swabs (Teigland type or other) |
| Sterile culture swabs (Culturettes) |
| Amies transport medium with charcoal |
| Viral transport media |
| Glass slides and coverslips |
| Sterile proctoscope (Welch Allyn, human pediatric type) or other endoscope, flexible or rigid |
| Sterile offset biopsy punch |
The smears may be fixed immediately in 95% alcohol, sprayed with a commercial fixative or hair spray, or left to dry in air.

A drop of new methylene blue stain placed on a coverslip and inverted on the smear can be used to examine a wet mount preparation immediately. This stain is not permanent and precipitates when it dries, and new methylene blue–stained smears cannot be used for comparison with other smears made later in the cycle. The Diff-Quik or Leukostat stain is a permanent stain that can be submitted for review by a pathologist. Examine the smear for stage of estrous cycle and evidence of active inflammation. Compare these findings with culture results and vaginoscopic findings to interpret evidence for an active genital tract infection, a carrier state of a potential infectious agent, or a possible contaminant at culture. A diagnostic laboratory with the ability to isolate specific infectious agents should indicate the number of organisms (few, moderate, many, or heavy) and report whether the isolates are pure or mixed and their significance.

**Examination of the Vagina**

The vagina of the bitch is long in comparison with that of other domestic animals, hence digital examination of the cervix, and in many cases the urethral orifice, is not feasible. The mucosa forms longitudinal folds. The clitoris is in a well-developed fossa in the floor of the vestibule. The vagina can be visualized completely with a small, sterile proctoscope or flexible endoscope. Lubricate the warmed, sterile instrument, and pass it to the region of the cervix. Examine first without insufflation for true color and vaginal fluids or discharge. When insufflation is performed while the vulva is compressed around the sterile proctoscope, the vagina expands and its entire wall can be viewed completely as the instrument is withdrawn.

The normal canine vagina has a uniform light pink color and longitudinal folds. During proestrus and estrus, the folds become more prominent and cross-striations give the surface a cobblestone appearance. This cobblestone appearance remains smooth when estrogen levels are high but quickly becomes angular (cobblestone appearance) when estrogen levels drop during the luteinizing hormone peak (ovulation), and progesterone levels increase. This change can be used to indicate ovulation and the ideal time for breeding. The hyperemia causes the vagina to appear reddish and congested. The pressure of air insufflation balances the mucosa. The canine vulva has a large cranial dorsal median fold that may obscure the cervix. In fact, ridges near the dorsal fold may give a false impression that this fold is the cervix. During estrogen stimulation, the cervix may be open and uterine blood may be escaping. In the management of dystocia, the vaginoscope can be used to detect puppies in the birth canal and to diagnose malpositions and aid in the correction of these conditions.

During the endoscopic examination, small tumors or polyps can be removed or large masses can be sampled with the biopsy punch. Ulcers or erosions can be cauterized, and foreign bodies can be removed.

A complete vaginal examination must include careful palpation of the vaginal wall and pelvic canal. This palpation is accomplished by digital examination through the vulva (using a sterile glove) and is assisted by palpation through the posterior abdominal wall. Incomplete hymen rings, vaginal fibrous stenotic rings, or pelvic malformation can be diagnosed. A digital rectal examination may be needed for vaginal masses or pelvic deformities.

**Estrous Cycle: Staging and Cytologic Findings—Canine**

The canine reproductive cycle begins at the age of 6 to 12 months and repeats at intervals of 4 to 12 months. In the average bitch, ovulation occurs spontaneously 1 to 3 days after the onset of estrus; in normal bitches ovulation may occur 3 days before to 11 days after the onset of estrus. Sperm live in the uterus of the estrous bitch up to 11 days, and the ovum lives up to 5 days after ovulation. The fertilized ovum takes 4 to 10 days to reach the uterus, and implantation takes place 18 to 20 days after ovulation. The gestation period from the first breeding is 57 to 72 days and from the luteinizing hormone peak is 64 to 66 days.
Anestrus
Anestrus is characterized by dryness of the mucosa and a thin vaginal wall with stratified squamous epithelial cells a few cells to several layers thick but without cornification. Noncornified epithelial cells and WBCs are present in a ratio of 1:5 in the vaginal smear. The WBCs are polymorphonuclear. The noncornified epithelial cells are 15 to 51 nm in diameter and have round free edges, granular cytoplasm, and large nuclei with distinct chromatin granules. The period of anestrus is 2 to 3 months or longer in some breeds.

Proestrus
In proestrus the vaginal wall is thicker than in anestrus, and the mucosa shows prominent cornified squamous epithelium (20 to 30 cells thick) with rete pegs. The longitudinal and transverse vaginal folds are thick, smooth, and round. The vaginal wall becomes impervious to WBCs, but there is extravasation of RBCs to the surface epithelium. The RBCs are discharged. Vaginal smears show predominantly RBCs and noncornified epithelial cells, which become cornified as proestrus progresses. WBCs are present, but their numbers decrease as estrus approaches. Debris and bacteria are abundant for 7 to 10 days.

Estrus
The vagina is thick with longitudinal and transverse folds that become angular as estrogen levels decrease and progesterone levels increase. Fluid is abundant, often tinged with blood. Noncornified epithelial cells and WBCs are absent. Cornified epithelial cells, which are polygonal and contain pyknotic nuclei or no nuclei, are predominant; their presence seems to be related to the appearance of flirting by the bitch and acceptance of the stud. WBCs reappear about 36 to 96 hours after ovulation. Bacteria and debris are absent during estrus, but they are seen again in the smears after ovulation when WBCs reappear 7 to 10 days later.

Diestrus
The number of WBCs increases rapidly, the number of cornified epithelial cells decreases, and the number of noncornified epithelial cells increases. After 5 to 7 days, the number of WBCs may decrease to 10 to 30 per field.

After parturition, much cellular debris, WBCs, RBCs, and a few epithelial cells are present for several days, until placental sloughing is complete. The presence of masses of degenerate WBCs (and bacteria) indicates metritis or endometritis. The continued presence of blood-tinged fluids containing abundant RBCs, a few noncornified epithelial cells, and occasional WBCs (nontoxic) plus necrotic cells for months postpartum is evidence of subinvolution of placental sites.

Estrous Cycle: Staging and Cytologic Findings—Feline
Most of the characteristics just discussed that apply to bitches also pertain to queens. However, the small size of the feline vagina precludes palpation and early vaginoscopy. A sterile, warm, small-animal otoscope speculum enables fairly good visualization of the vaginal mucosa and can be used with a small, 4-mm–diameter sterile swab to obtain smears for culture procedures. Use of the speculum is easiest after parturition or during estrus.

Vaginal cells for cytologic examination can be obtained with a moistened 3-mm cotton swab (Calgiswab) inserted 2 cm into the vagina. In some cases, flushing the vagina with sterile saline injected and aspirated with a clean glass eyedropper is more successful. Use of an eyedropper may trigger ovulation, as it simulates coitus.

Unlike the bitch, the queen shows no diapedesis of RBCs during proestrus or throughout the estrous cycle. Cytologic examination of feline vaginal smears reveals the following by stage of the estrous cycle.

Anestrus or Prepuberty
Cytologic examination reveals scarce debris and numerous small, round epithelial cells with a high nuclear/cytoplasmic ratio, frequently in groups (seasonal: from September to January in the Northern Hemisphere).
**Proestrus**
Cytologic examination reveals increased debris and fewer but larger nucleated epithelial cells with a low nuclear/cytoplasmic ratio (0 to 2 days).

**Estrus**
Cytologic examination reveals markedly less debris and numerous large polyhedral cornified cells with curled edges and small dark pyknotic nuclei or loss of nuclei (6 to 8 days) after coitus or induced ovulation.

**Early Diestrus**
Cytologic examination reveals hazy, ragged-edged cornified cells and zero to numerous WBCs with numerous bacteria and increased debris.

**Late Diestrus**
Cytologic examination reveals increasing numbers of small basophilic cells with WBCs still present (total period of metestrus, 7 to 21 days). If ovulation does not occur, the smear will return to an anestrous stage with few to no WBCs.

The feline estrous cycle is continuous every 14 to 36 days if 12 to 14 hours of light are present daily. Ovulation is induced 24 to 30 hours after coitus. Sperm require 2 to 24 hours for capacitation in the uterus. Implantation is expected 13 to 14 days after coitus.

**Additional Reading**
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Grundy SA, Davidson AP: Feline reproduction. In Ettinger SJ, Feldman EC, editors: *Textbook of veterinary internal medicine*, ed 6, St Louis, 2005, Elsevier.
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**Artificial Insemination: Canine**
The procedure for artificial insemination in dogs includes the following steps:

1. Determine the correct time to inseminate by test-teasing with a stud, by cytologic examination of vaginal smears, or by vaginoscopic examination to determine the day when vaginal folds change from round to angular. Breed the day after the bitch first stands staunchly to accept service and “flags” her tail or during cytologic indications of estrus (complete cornification of vaginal epithelial cells) but before WBCs reappear in the smears. Breed at 48-hour intervals until the female dog goes out of heat or for three or four inseminations.

2. If the vulva is soiled, clean it thoroughly with alcohol swabs (Box 4-15).

3. Gently aspirate semen through the inseminating pipette into the warm syringe.

**Box 4-15 Materials Used for Performing Artificial Insemination**
- Dry, warm, sterile 5- or 10-mL syringes
- Rubber adapter tubing, ¾-inch long
- A 6- to 9-inch plastic or polypropylene inseminating pipette
- A sterile examination glove
- Alcohol
- Cotton
Do not use lubricating materials.
4. Using a gloved left index finger (not lubricated) as a guide, insert the pipette through the vulva and dorsally into the vagina and forward to the cervix. Elevate the bitch's rear quarters to a 45-degree angle by having an assistant pick up the bitch by holding the hock region so that no pressure is applied to the ventral abdomen and uterus. Eject the semen gently and slowly. Eject a bubble of air to push all the semen through the pipette. Deposit the semen in the anterior vagina.

5. Remove the pipette, and hold the bitch in an elevated position for 5 minutes. During this time, use the finger encased in a sterile glove to "feather" the ceiling of the vagina to stimulate constrictor activity. This may be important to simulate a "tie" and transport semen into the uterus.

6. Lower the bitch to the normal position, and immediately walk her for 5 minutes so that she does not sit down or jump up on a person and allow semen to run back out of the vagina.

7. For best conception, inseminate undiluted fresh semen immediately.

8. Refrigerated extended semen is best used within 24 to 48 hours if possible. However, refrigerated semen has been kept viable for up to 9 days with proper care. Skim milk has been used as an economical and adequate extender. Heat milk to 92° to 94° C for 10 minutes, cool it, and skim it at room temperature. To each milliliter, add 1000 units of crystalline penicillin. If Pseudomonas species affect the semen, polymyxin B may be added at 200 units/mL of extender. Dilute semen with extender at a semen/extender ratio of 1:1 to 1:4. Extend canine semen for freezing with a diluent containing 11% lactose, 4% glycerin, and 20% egg yolk. Refrigerate the 1:4 diluted semen; then pipette 0.05-mL portions into depressions in a block of dry ice and hold them for 8 minutes to freeze. Store the frozen pellets in liquid nitrogen. Frozen semen can be thawed in buffered saline at 30° to 37° C. Good semen may be stored in liquid nitrogen for many years without significant loss of motility. Conception is best when large numbers of thawed motile sperm are deposited in the cervix or uterine cavity. Conception is poor when thawed semen is placed in the anterior vagina, as done in artificial breeding with raw semen.

Additional Reading
Memon MA, Sirinarumitr K: Semen evaluation, canine male infertility, and common disorders of the male. In Ettinger SJ, Feldman EC, editors: Textbook of veterinary internal medicine, ed 6, St Louis, 2005, Elsevier.

REPRODUCTIVE TRACT: MALE

Semen Collection: Canine

Semen is collected for examination for breeding soundness, for investigation of infertility or prostatic disease, and for artificial insemination (Box 4-16).

The following steps outline the procedure for collecting semen from a male dog:

1. Take the stud and an estrous teaser bitch (if available) to a quiet room where there will be no distractions and where there is good traction (rubber mats or rug) for mounting by the stud.

2. Hold the bitch, and allow the stud to “flirt” (become aroused) for several minutes. If the bitch is in heat, a brief period of foreplay (“foreplay” may not be the appropriate word to describe the mating behavior of dogs, but it illustrates the point) with both dogs unrestricted will help the process.

3. If necessary, have assistants restrain the muzzled bitch and control the stud by a collar and leash. Bring the stud up to the rear end of the bitch, and allow him to mount her or keep his nose in the region of her perineal area.

4. Attach the artificial vagina to the semen collection tube, and apply a scant amount of lubricant to the opening of the artificial vagina.

5. If mounting occurs, allow the stud to grasp the bitch and start to thrust his pelvis in an attempt to copulate. Gently, from the side of the sheath, grasp the penis by the prepuce and move the prepuce back over the engorged bulbus glandis; while
applying the artificial vagina to the shaft of the penis, apply pressure with the thumb and forefinger proximal to the exposed glandis. This usually can be done with one motion as the stud is thrusting. If the stud is shy and not interested, massage the penis slightly in the prepuce or in the artificial vagina to cause erection. When erection of the bulbis is felt, reflect the prepuce posteriorly to free the bulbis. Apply pressure with the thumb and forefinger behind the bulbis, circling the shaft of the penis. After completion of the most rapid pelvic thrusting and ejaculation of the sperm-rich fractions of semen (1 to 3 mL), twist the penis 180 degrees backward in a horizontal plane, between the hind legs, so that the penis remains in the same plane as in the forward position, with the thumb and forefinger still applying pressure around the circumference of the penis proximal to the bulbis. The penis cannot be twisted unless the prepuce is reflected posterior or proximal to the bulbis glandis. Twisting the penis in this position simulates a natural “tie” and allows the person collecting the semen to better visualize the collection (artificial vaginas are widely available now and are much preferred because they simulate the natural pressure of the vagina). The first drops of ejaculate may be discarded, especially if any urine is present. Collect the sperm-rich fraction separately. A clear ejaculate is prostatic fluid, which may be collected separately for examination.

6. After semen collection, place the penis in the forward position, straighten out the prepuce to avoid paraphimosis, and remove the bitch from the room. Allow the stud to lick the erect penis and lose the erection. Check the stud for evidence of paraphimosis before it is released or caged. The ejaculate consists of three fractions:

- **First fraction**: Urethral secretion (usually clear fluid)—0.1 to 2 mL within 50 seconds, pH 6.3. If evidence of urine is present, discard this fraction and do not add it to the sperm-rich fraction. In most ejaculates collected from dogs, the first and second semen fractions are collected together.

- **Second fraction**: Sperm-containing secretion (milky opaque fluid)—0.5 to 3 mL within 1 to 2 minutes, pH 6.1.

- **Third fraction**: Prostatic secretion (usually clear fluid)—2 to 20 mL within 30 minutes, pH 6.5. The total specimen is 0.3 to 20 mL, pH 6.4. Because the first and third fractions are clear, waterlike material and the second fraction is milky-opaque, the clinician can separate them by changing collecting tubes as each fraction is ejaculated. Collection of only enough prostatic fluid to rinse the sperm fraction into the test tube is best. Too much prostatic fluid may be detrimental to the longevity of sperm in storage. Collecting individual fractions may be important in determining the site of an inflammatory reaction, but for artificial insemination only the sperm-rich, low-volume ejaculate is needed for insemination, dilution, or freezing.

7. Return the stud to his cage. Retain the bitch until the semen is examined, if insemination is to be performed.
Evaluation of Semen

Immediately after semen collection, slowly invert the tube several times to mix the semen gently. Determine the motility of sperm by placing one drop of semen on a warmed microscope slide. Cover the slide with a coverslip, and observe the specimen under low power for progressive motility. There will be no "waves," but general vigorous forward motion should be evident. If the sample is too concentrated for individual sperm to be found, mix one drop of semen with one drop of saline at body temperature on a warmed microscope slide. Using high power, count 10 different groups of 10 sperm, observing the numbers of motile and nonmotile sperm. Total motility for a suitable sample should be 80% or greater. Motility less than 60% is not satisfactory.

Determine the number of sperm in the total ejaculate. Sperm concentration may be determined in a hemocytometer with a 1:100 blood cell dilutor kit (Unopette), and concentration then is multiplied by volume to determine sperm numbers per ejaculate. Remember that more dilute samples will be obtained when prostatic fluid is collected, but total sperm numbers in the ejaculate will be only marginally influenced by dilution with prostatic fluid. Total sperm per ejaculate should exceed 300 million in a normal male dog and may approach 2 billion in large dogs. A minimum number of 200 million sperm per insemination is needed on average for conception.

Determine morphology. Make a smear of a drop of semen like a blood smear and allow it to air-dry. Then stain the smear with Diff-Quik stain; dip the slide into the fixative and solutions 1 and 2 for 2 to 3 minutes each. Then count 100 sperm at ×1000 magnification, noting normal and abnormal sperm. If there is any question about abnormality, examine 500 sperm cells.

Normal canine sperm are 63 nm long; the heads are 7 nm long. The percentage of abnormal sperm should be less than 20%. Differential abnormality is important, and the following abnormalities should not be exceeded in any sperm count: abnormality of the head, 10% to 12%; midpiece abnormalities, 3% to 4%; tail abnormalities, 3% to 4%; and retained protoplasmic droplets, 3% to 4%. Figure 4-5 shows abnormalities that should be counted and recorded. The presence and location of distal or proximal protoplasmic droplets, which may indicate cell immaturity, are important to note.

Defects of the cells within the testes are generally more serious than defects that occur in the sperm during epididymal transport or after ejaculation (such as fractured heads, retained protoplasmic droplets, or bent tails). Usually a biopsy should not be done on material from testes unless the testes are azoospermic. Damage produced after the sperm have left the testes may indicate epididymal disease or may be the result of cold, trauma, or osmotic or urinary contamination. When abnormalities are found, it is wise to obtain two or three semen samples within a few days for baseline evaluation and then repeat the studies in 4 to 6 weeks to determine whether there is a healing or regressing trend. There are usually 64 days from the date of sperm formation to the date of ejaculation: 54 days in the testes and 10 days in transport and maturation in the epididymis.

Normal male dogs can be used at stud once every other day indefinitely or once every day for 7 to 9 days, after which sperm numbers in the ejaculate will decline but not to less than the numbers needed to achieve conception.

Additional Reading

Baker R, Lumsden JH: The reproductive tract: vagina, uterus, prostate, and testicle. In Baker R, Lumsden JH, editors: Color atlas of the cytology of the dog and cat, St Louis, 2000, Mosby. (Note: This textbook contains exceptional color plates of normal and abnormal reproductive tract cytologic findings of the dog and cat.)

Feldman EC, Nelson RW: Canine and feline endocrinology and reproduction, ed 3, St Louis, 2004, Elsevier.

Memon MA, Sirinarumitr K: Semen evaluation, canine male infertility, and common disorders of the male. In Ettinger SJ, Feldman EC, editors: Textbook of veterinary internal medicine, ed 6, St Louis, 2005, Elsevier.

Wright PJ, Parry BW: Cytology of the canine reproductive system, Vet Clin North Am Small Anim Pract 19:851–874, 1989.
Although castration is a common first recommendation for any male dog with known or suspected prostatic disease, a number of prostatic disorders are recognized for which cytopathologic and histopathologic examination, rather than castration, is indicated. Benign prostatic hyperplasia is recognized as the most common prostatic disorder of male dogs. In half of the dog population, changes consistent with benign prostatic hyperplasia are present by 4 to 5 years of age, especially in older intact dogs. Because benign prostatic hyperplasia is androgen
dependent, routine castration is the recommended treatment. However, at least three differential diagnoses justify additional diagnostic tests: prostatic neoplasia (usually adenocarcinoma), acute and chronic bacterial prostatitis, and prostatic cysts (septic and nonseptic).

In male dogs with prostatomegaly and associated signs (dysuria and/or dyschezia), further evaluation of the prostate is indicated. Several techniques have been described. Abdominal ultrasonography is the preferred technique for evaluating prostate size, shape, and consistency. Distension retrograde contrast urethrocystogram has been described as a means for evaluating the internal integrity of the prostate and is moderately effective in distinguishing normal from abnormal. However, this technique is not known to distinguish among various types of prostate disease.

**Patient Preparation**

Cytologic examination and quantitative bacterial culture of the ejaculate (especially the third fraction) of a male dog is recommended in any patient with prostatomegaly. However, sample collection can be difficult and is frequently not successful. In addition to lumbar radiographs and abdominal ultrasonography, performing a prostatic wash is a simple, noninvasive technique that may yield diagnostic information.

**Technique**

Using aseptic technique, place a conventional urinary catheter into the bladder and remove all urine. Lavage of the urinary bladder with up to 5 mL of sterile saline is recommended. Recover the saline and save it (sample No. 1). Subsequently, retract the catheter tip, but only to the level of the prostate gland (immediately caudal to the trigone). Position of the tip usually can be verified by tactile placement and the detection of increased resistance to catheter movement during retraction. Position can be confirmed with a lateral radiograph of the pelvis.

With the catheter in place, identify the prostate on a digital rectal examination and gently massage for approximately 1 minute to force prostatic fluids into the urethra. Infuse 5 mL of sterile saline through the catheter. The objective is to wash prostatic fluids and cells into the urinary bladder and recover the saline from the bladder (sample No. 2).

Examine fluid from both samples cytologically by distributing a drop of fluid across a glass slide, air-drying, and staining; submit a small aliquot (0.5 mL) for bacterial culture. Cytologic examination is used to detect the presence of inflammatory cells versus neoplastic cells. Low numbers of neutrophils (<5 cells per high-power field) are present in ejaculates and prostatic washes from normal dogs. Quantitative bacterial culture, with a yield of greater than 2 log_{10} of one or more bacterial species in sample No. 2 confirms bacterial prostatitis.

**Special Considerations**

Complications from this procedure are unlikely, but conceivably a patient with septic prostatitis and prostatic abscesses could become bacteremic after this procedure, which in some patients could lead to sepsis.

**Prostate Biopsy and Fine-Needle Aspiration**

**Patient Preparation**

Ultrasound examination is an important first step, when available, in assessing the size, shape, and internal integrity of the canine prostate gland and for detecting any changes in structures adjacent to the prostate. However, ultrasonography generally will not distinguish among different types of prostatic disease. Further diagnostic tests are especially indicated in castrated, middle-aged to older male dogs with evidence of prostatomegaly. Percutaneous fine-needle aspiration and/or prostatic biopsy are indicated.

**Technique**

Fine-needle aspiration of the prostate is performed through a ventral abdominal approach. Use aseptic technique, and surgically prepare the skin at the level of needle insertion. Because needle movement, once the needle has been inserted, could damage the urethra or
adjacent structures, perform the procedure in the sedated or anesthetized patient. Use an approach similar to that used for cystocentesis in a male dog with the exception that needle entry is at a point caudal to that used to enter the urinary bladder but is cranial to the pubis. The procedure can be performed with or without ultrasound guidance. In the absence of ultrasound guidance, determine needle position by tactile placement and detection of resistance as the needle enters the prostate. Multiple needle penetrations and aspirations are attempted without withdrawing the needle from the skin. Relieve negative pressure in the syringe before removing the needle. Apply any material collected to a glass slide and allow it to air-dry before staining. Any conventional stain used for peripheral blood is appropriate.

A transrectal approach to fine-needle aspiration of the prostate has been used in dogs and is performed routinely in men. However, the distance from the anus to the prostate, visualization, and the risk of infection generally are cited as reasons for not performing this technique in dogs.

Fine-needle aspiration may not be diagnostic, particularly in patients with isolated, discrete lesions (cysts or neoplastic nodules) within the prostatic parenchyma. In such cases, ultrasound-guided needle (Tru-Cut) biopsy of the prostate is indicated. Specific training and experience are indicated for performing this procedure because significant complications can result.

Special Considerations
Complications associated with prostate biopsy and fine-needle aspiration are not insignificant. Hematuria and periprostatic hemorrhage are described. Postaspiration and postbiopsy abscess also have been described. Consider the risk of urethral penetration and subsequent stricture at the site of penetration.

Additional Reading
Kutzler MA, Yeager A: Prostatic diseases. In Ettinger SJ, Feldman EC, editors: Textbook of veterinary internal medicine, ed 6, St Louis, 2005, Elsevier.

**RESPIRATORY TRACT PROCEDURES**

**Upper Respiratory Tract**
For purposes of this discussion, the anatomic limits of the upper respiratory tract of the dog and cat extend caudally from the nasal planum to the first tracheal ring. Key anatomic structures that principally can cause clinical signs include the anterior (external) nares, nasal cavity, nasal turbinates, frontal sinuses, maxillary recesses, upper dental arcade (especially the roots of the maxillary canine teeth), choanae (posterior nares), nasopharynx, soft palate, arytenoid cartilages, glottis, larynx, and vocal folds (see Table 4-9).

| **TABLE 4-9** Anatomic Limits of the Upper Respiratory Tract and Defining Clinical Signs |
|--------------------------------------------------|-------------------------------------------------|--------------------------------------------------|
| **Compartment**                                  | **Anatomic Limits**                             | **Defining Clinical Sign(s)**                    |
| I                                                | Nose, nasal cavity, and paranasal sinuses      | Sneezing and/or nasal discharge                  |
| II                                               | Nasopharynx, posterior nares (choanae), and    | Stertor (snort) and reverse sneeze              |
|                                                  | soft palate                                     |                                                  |
| III                                              | Larynx                                         | Stridor (wheeze)                                |
Clinical signs related to the upper respiratory tract in dogs and cats are among the most common presenting complaints encountered in small animal practice and, interestingly, are frequent reasons for referral to specialty practices and veterinary teaching hospitals. The oral and nasal cavities are important portals of entry for foreign body entrapment and infectious agents. In addition to the occurrence of nasal neoplasia and trauma, it is not surprising that upper respiratory tract diseases in dogs and cats are common presentations. However, upper respiratory signs can be associated with significantly different underlying causes. Localizing the problem amid a variety of clinical signs in an anatomically complex area presents significant diagnostic and therapeutic challenges to even the most astute clinician. The presentation addresses upper respiratory disease in the dog, with specific emphasis on clearly defining the presenting clinical signs, localizing the problem, and establishing the diagnosis.

Clinical Signs
The first and most important step in establishing a diagnosis of canine upper respiratory disease is to define the presenting sign. Experience has shown that an owner's ability to describe the patient's clinical signs accurately, particularly when signs are not present at the time of examination, is usually inconsistent and inaccurate, although it can be most entertaining. The four localizing clinical signs characteristically associated with upper respiratory diseases are sneezing and/or nasal discharge, stertor, stridor, and cough. Each sign, considered independently, will focus the examination to the appropriate anatomic region of the upper respiratory tract.

Sneezing and/or Nasal Discharge
Definition of the clinical signs sneezing and nasal discharge may seem intuitive. This is the most common presenting sign in dogs with upper respiratory disease. Owners that present a dog with sneezing are likely to be accurate in their description of the problem. However, the presence or absence of a nasal discharge may be more difficult to establish. Volume, character, and frequency of the discharge ultimately determine whether the owner will have even observed this sign. The astute owner will report whether the discharge is unilateral or bilateral. In the patient that has a history of sneezing and nasal discharge, instillation of a topical nasal decongestant into each nostril occasionally will provoke sneezing and elicit the nature of any discharge that is present.

Sneezing and/or nasal discharge localizes the problem to the nose, nasal cavity, and paranasal sinuses. However, thorough examination of the nose and nasal cavity can be difficult, even with the availability of appropriate endoscopy equipment. In addition to careful examination of facial symmetry, the first part of the examination begins in the oral cavity, with emphasis on the maxilla, the hard palate, and the canine teeth. Examine the hard palate for evidence of trauma (penetrating or nonpenetrating) and congenital cleft palate (puppies). Carefully probe the medial aspect of the maxillary canine teeth for evidence of oronasal fistulas. Despite normal-appearing teeth and gingiva, severe, occult periodontal disease with resulting necrosis of bone does result in a septic communication between the oral and nasal cavities. The owner characteristically describes paroxysms of sneezing associated with a sanguineous nasal discharge or spray.

If these findings are negative, radiographs of the skull are indicated. Three views, obtained in the anesthetized patient, are indicated: lateral, ventrodorsal, and occlusal (open mouth) view. Radiographic interpretation of the nasal cavity and sinuses dictates that the clinician have a thorough understanding of the anatomy of the upper respiratory tract. Subsequently, with the patient still anesthetized, attempt a visual examination of the nasal cavity. Radiographs are always performed before visual examination of the nasal cavity. Manipulation of the tissue may result in intranasal bleeding, which will significantly complicate radiographic interpretation. A simple otoscope speculum placed into each nostril allows an adequate examination of the proximal 20% to 25% of the nasal cavity in most dogs. Visual examination of the caudal 75% of the nasal cavity can be attempted only with a small-diameter endoscope. Flexible and rigid scopes are available; each has advantages and disadvantages that will be discussed. Computed tomography and magnetic resonance...
imaging are important alternative diagnostic tools; however, expense and lack of availability are significant limiting factors.

Understanding the most commonly diagnosed causes of sneezing and nasal discharge is especially helpful in patient management. In no particular order, the most common differential diagnoses for sneezing and/or nasal discharge include the following:

1. **Oronasal fistulas**: Especially common in middle-aged to older dogs, despite a history of recent dental prophylaxis. Empiric treatment with an orally administered antibiotic typically results in rapid and complete resolution of clinical signs, but only during the time the patient is receiving the antibiotic. Diagnosis is confirmed by probing the gingival sulcus of the upper canine teeth.

2. **Nasal neoplasia**: Most commonly reported in dogs 8 to 10 years of age (range: 1 to 15 years of age). No breed is predisposed, but the condition is uncommon in brachycephalic breeds. Persistent nasal discharge, sneezing, and intermittent epistaxis are common presenting signs. Nasal radiographs may demonstrate lytic bone lesions. Lysis of the vomer strongly supports neoplasia versus mycotic rhinitis. Exposure to tobacco smoke has been associated with 2.5 times greater risk in long-nosed dogs. No or minimal response of the discharge to antibiotics occurs. Eighty percent of nasal tumors are malignant. Adenocarcinoma is most common, followed by squamous cell carcinoma. Sarcomas account for small number of nasal tumors.

3. **Mycotic rhinitis**: Difficult to distinguish from neoplasia. Persistent and voluminous mucoid nasal discharge, with or without sneezing, and nasal pain are reported. Erosion of the external nares is an important physical finding. Discharge is not responsive to antimicrobial treatment. Occlusal view radiographs of the nasal cavity may demonstrate evidence of turbinate destruction and/or increased fluid density on the affected side. Forty percent of patients are 3 years of age or younger; 80% are 7 years of age or younger. The diagnosis is uncommon in brachycephalic breeds. Localized *Aspergillus fumigatus* infection is reported most commonly.

4. **Lymphoplasmacytic rhinitis**: Poorly described clinical syndrome associated with chronic sneezing and nasal discharge (bilateral or unilateral). Affected dogs are typically young to middle-aged, large-breed dogs. Signs are not usually responsive to antibiotics or steroids (topical or systemic). Diagnosis is based on ruling out other causes and nasal biopsy findings.

**Stertor**

The second most common clinical sign associated with upper respiratory disease in dogs, stertor is intermittent, yet persistent or continuous snorting, also called stertorous breathing. Paroxysms of stertor, typically called reverse sneezing, are characterized by rapid, consecutive inspiratory bursts through the nose. Seldom actually seen during examination, reverse sneezing is likely to result from the patient's attempt to displace matter trapped in the nasopharynx and move it into the oropharynx, where it can be swallowed.

Visualization of the nasopharynx and choanae is essential in the patient that has chronic or persistent stertor. The examination can be accomplished only in the anesthetized patient. Sedation is not sufficient to conduct the examination. A flexible endoscope with the ability to flex approximately 170 to 180 degrees is recommended. Examination allows visualization of the nasopharynx and associated mucosa, the choanae (posterior nares), and the top of the soft palate (see Figure 4-34).

Nasopharyngeal foreign bodies are by far the most common finding. Sticks, plant material (grass and juniper twigs), peas, cotton balls, and thread are just a few examples. Neoplasia is the second most common finding. In cats, lymphoma (FeLV related) obstructing the choana most commonly is observed (see Figure 4-35). In dogs, neoplasia is uncommon, but (in my experience) sarcomas in young dogs have been seen most frequently.
**Stridor**

The least commonly encountered of the upper respiratory signs is stridor, or stridulous breathing. Stridor is audible wheezing and is associated with restriction to airflow, usually at the level of the larynx. Therefore stridor is the most critical and potentially life-threatening upper respiratory sign. This is especially true when stridor is continuous. The patient that has continuous stridor deserves immediate attention. Make every effort to discern the cause once the clinical sign is characterized. In obtaining the history, owners generally describe wheezing accurately; however, some patients actually may have severe dyspnea or orthopnea. Careful questioning of the client is indicated to determine whether wheezing is associated with the additional effort to breath. The clinician also should make an effort to discern whether the owner has observed any change in the ability of the dog to vocalize or bark.

Simply listening to the patient breath in a quiet room is the first step in assessing stridor. A stethoscope is not required to hear wheezing but should always be used to examine the cervical trachea, the larynx, and the lungs. Any restriction to airflow in the larynx or cervical trachea can cause stridor. However, in the majority of cases the stridor will be significantly louder at the level of the larynx, indicating a restrictive lesion at that level.

If any indication of respiratory distress is reported or manifests during the examination, subject the patient to a visual examination under general anesthesia. Sedation is not sufficient to conduct the examination. Be prepared. These patients are not routine. Emergency resuscitation may be required on induction of anesthesia, including the need to perform a tracheostomy.

On induction, carefully place an endotracheal tube. If there are no complications associated with inserting the tube, once anesthesia has been effectively induced and the patient’s condition is stable, lateral and dorsoventral radiographs of the larynx and cervical trachea are indicated. Metallic objects (e.g., fish hooks) can become buried in the mucosa and may not be observed during a visual examination.

Remove the endotracheal tube in order to conduct a visual examination. A focal, hands-free light source directed into the oropharynx is strongly recommended. Carefully examine the epiglottis, arytenoid cartilages, glottis, and vocal folds using a cotton-tipped applicator. Careful observation of the symmetry and function of the arytenoid cartilages is essential. The left and right cartilages normally respond to tactile stimuli when the patient is in a light plane of anesthesia; both sides should move to the medial plane rapidly and at the same time. They may not close, depending on the depth of anesthesia. It should be possible to visualize the cartilage on the inside of the tracheal rings while looking through the glottis.

In large breed, middle-aged and older dogs, laryngeal paralysis is the most common cause of stridor. Associated signs may include exercise intolerance and collapse during exertion. Laryngeal paralysis and stridor also may be observed in young breeds as a congenital disorder (Dalmatian, Rottweiler, Bouvier des Flandres, Siberian Husky, and Bull Terrier). Foreign body penetration of the laryngeal tissues can cause serious and life-threatening obstruction because of infection and swelling. Neoplasia may cause obstructive mass lesions involving the larynx, especially squamous cell carcinoma and lymphoma. Granulomatous laryngeal disease and fungal mycetoma have been reported.

The presence of a mass lesion, assuming there is no foreign body detected, warrants biopsy of the lesion. Additional effort to control postbiopsy bleeding is important. I use a cotton-tipped applicator saturated with a 1:10,000 dilution of epinephrine held against the biopsy site for 30 to 60 seconds. This is time well spent. Postbiopsy administration of systemically effective dexamethasone has been suggested to control laryngeal swelling, but I have not found this to be effective or important.

**Additional Reading**

Holt DE: Upper airway obstruction, stertor, and stridor. In King LG, editor: *Textbook of respiratory disease in dogs and cats*, St Louis, 2004, Elsevier.

Van Pelt DV, Lappin MR: Pathogenesis and treatment of feline rhinitis, *Vet Clin North Am Small Anim Pract* 24:807, 1994.
Lower Respiratory Tract

The following diagnostic procedures are elective and are indicated in patients with chronic disorders of the lower respiratory tract that are not considered life-threatening.

Transtracheal Aspiration

Transtracheal aspiration is a safe and clinically useful method for obtaining material for cytologic and bacteriologic examination from the lower respiratory tract of medium-sized to large dogs without invading the oval cavity. This procedure is not indicated in cats.

**Patient Preparation**

The technique can be performed on the unanesthetized animal, although some sedation may be indicated. The hair overlying the larynx is clipped and prepared surgically. In small dogs and cats, tracheal aspirates are collected by passing the catheter through sterile tracheal tubes. Light levels of anesthesia are used to accommodate coughing and tracheal intubation.

**Technique**

Place the animal in sternal recumbency or in the sitting position. Elevate and extend the head. Locate the cricothyroid membrane by moving the finger along the proximal trachea until the large ventral ridge of the cricoid cartilage is felt. Use a 16-gauge, ½-inch intravenous catheter to collect material through the trachea (Figure 4-52). Puncture the cricothyroid membrane with the 16-gauge needle, and pass the catheter into the trachea until it reaches the distal trachea or main stem bronchus. (Alternatively, in large dogs, insert the catheter between the tracheal rings at the junction of the middle third and distal third of the cervical trachea.) Withdraw the needle, and leave the catheter in place. Attach a 12-mL syringe containing sterile saline solution to the catheter. Expel 1 to 2 mL of saline from the syringe. When the animal coughs, aspirate with the syringe to collect cells and mucus for bacteriologic and cytologic examination. When material has been collected, remove the catheter and bandage the animal’s neck. Culture material present in the syringe in blood agar and in thioglycolate medium. Prepare material from aspiration for cytologic examination. Press large plugs of mucus between two clean glass slides, and stain thin smears with Wright or Giemsa stain.

**Special Considerations**

Complications of transtracheal aspiration biopsy include catheter trauma to the lower airway or needle trauma to the larynx, resulting in bleeding, subcutaneous emphysema, pneumomediastinum, pneumothorax, or airway obstruction.

Endotracheal Wash

In cats and small dogs and in dogs for which general anesthesia is not contraindicated, tracheal aspiration (or tracheal wash) is a relatively safe, easy-to-perform procedure that can yield excellent diagnostic cytologic and culture specimens. The procedure has some advantages over transtracheal aspiration in that it allows sample collection from airways beyond
the bifurcation of the trachea (carina) and avoids complications associated with patient discomfort and movement during the procedure. However, cough reflexes are eliminated completely, thereby decreasing potential sample yields from deep in the airway structure. In either case, transtracheal and tracheal aspirations provide the best diagnostic material from large airways, not small airways and alveoli.

Figure 4-52:  A, Diagrammatic representation of anatomic structures involved with transtracheal aspiration technique. The best landmark for percutaneous puncture is the cricothyroid ligament of the larynx, although the tracheal lumen also can be entered between cervical tracheal rings. B, The needle is advanced and directed slightly caudal until the trachea is entered. Once the needle is positioned within the tracheal lumen, the catheter is advanced through the needle and down the trachea. (From Kirk RW: Current veterinary therapy VIII: Small animal practice, Philadelphia, 1983, WB Saunders.)
Patient Preparation
The anesthetized dog or cat usually is placed in sternal recumbency. Lateral recumbency (affected side down) may facilitate recovery of specimens from patients with focal or regional lung disease. Use a sterile endotracheal tube to administer the anesthetic and oxygen.

Technique
Introduce a sterile red rubber catheter (long enough to extend beyond the carina) through the endotracheal tube (Figure 4-53). (Note: Disposable adapters for use with endotracheal tubes are available that allow continuous administration of anesthetic gases while passing the rubber catheter through the tube [Figure 4-54].) Introduce the catheter blindly until resistance is met as the tube attempts to enter smaller airways.

Use aliquots of warmed, sterile saline in prepared syringes to wash and retrieve samples. Aliquots of 3 to 5 mL can be used per collection attempt in small dogs and cats, whereas volumes up to 10 and 20 mL are appropriate for larger dogs. With the catheter positioned as deep as practical in the airway, infuse the entire volume of saline. Gentle agitation (intermittent aspiration and injection) may facilitate sample collection. If a 10-mL quantity is infused, retrieval of only 1 to 2 mL as a final volume per collection attempt is not unusual. The remaining fluid is rapidly (seconds) absorbed into the pulmonary vasculature. Important: When performing this procedure, do not withdraw the rubber catheter while maintaining a high negative pressure on the syringe. Doing so actually may tear mucosa away from the airway and could lead to pneumothorax or pneumomediastinum.

Special Considerations
The procedure can be repeated safely in the same patient several times. Collection of three to five samples is routine. More samples may be indicated depending on the patient's condition and response to the procedure. Monitoring of patients undergoing a tracheal wash procedure for oxygen saturation (pulse oximetry) throughout the procedure is recommended. In some patients with reactive airways, infusion of saline may cause significant bronchoconstriction, detected by a rapid decline in oxygen saturation.

Process collected samples immediately. Submit at least one sample of liquid (not a swab of the liquid) for bacterial culture and sensitivity or MIC. Quantitative cultures are

Figure 4-53: Endotracheal wash performed directly through a prepositioned endotracheal tube in a cat.
impractical because specimens will be diluted. If the sample appears to be highly cellular (characterized by turbidity), place aliquots into tubes containing EDTA.

**Additional Reading**
Syring RS: Tracheal washes. In King LG, editor: *Textbook of respiratory disease in dogs and cats*, St Louis, 2004, Elsevier.

**Bronchoalveolar Lavage**
BAL is an alternative diagnostic procedure to transtracheal aspiration and endotracheal wash. BAL has the advantage of retrieving fluid samples from distal airways and alveoli. This is a highly diagnostic procedure indicated in patients with generalized lung and regional (interstitial and/or airway) disease that are not in respiratory distress. Patients suspected of having allergic or infectious respiratory disease or neoplasia are candidates for BAL. Although BAL is used as a therapeutic procedure in human beings with chronic lung disease associated with accumulations of surfactant in the alveoli, there is no therapeutic indication for BAL in dogs or cats.

**Patient Preparation**
BAL must be performed in the anesthetized patient and consequently may be contraindicated in some patients with severe respiratory disease.

**Technique**
BAL entails instilling sufficiently large volumes of fluid into the distal airways to reach, and recover, reasonable cytologic samples representative of small airways and alveoli. Several variations on the technique have been described, but all recommend blind or visual placement of a catheter or bronchoscope into an airway of a lung lobe such that the airway is occluded. Sterile, nonbacteriostatic 0.9% saline, warmed to approximately body temperature and drawn into prepared syringes, is the fluid of choice. The volume of fluid varies with the size of the patient. Defined doses of saline per kilogram of body mass have not been described. In large dogs, two 25-mL aliquots (50 mL total) can be infused into each lobe sampled. In small dogs and cats, total volumes per lobe generally are restricted to 10-mL aliquots. Recovery may be as low as 2 to 5 mL with each attempt.
For dogs undergoing BAL, particularly when reactive (allergic) airway disease is suspected, pretreatment with a bronchodilator is appropriate and is recommended. Aminophylline can be administered at 5 mg/kg (cats) or 11 mg/kg (dogs) orally 1 to 2 hours before the procedure. Alternatively, terbutaline, 0.01 mg/kg, can be administered subcutaneously to cats 30 minutes before the procedure.

Bronchoscopic BAL allows direct visualization of the airway or lobe of interest. In medium to large dogs, place the bronchoscope directly through a sterile endotracheal tube. Use of an inexpensive, disposable endotracheal tube adaptor permits simultaneous administration of oxygen and anesthetic throughout the procedure. Saline can be infused from a syringe directly through the biopsy channel of the endoscope. The bronchoscope serves as the infusion catheter. Using this technique, samples can be collected effectively from multiple lobes. Blind placement (nonbronchoscopic) BAL using a rubber end-hole catheter is required in cats and small dogs. Blind placement is also appropriately used in patients with generalized lung or airway disease when discrete placement of the bronchoscope cannot be accomplished reliably.

Special Considerations
As with the endotracheal wash procedure described before, gentle agitation with the syringe (intermittent aspiration and injection) may facilitate sample collection. Do not withdraw the bronchoscope or catheter while maintaining significant negative pressure because this may lacerate the airway, leading to pneumothorax or pneumomediastinum.

BAL is an invasive diagnostic procedure that is not without risk of injury or death. After completion of BAL, administration of 100% oxygen for 5 to 10 minutes via endotracheal tube is recommended for all patients. Evaluate the patient carefully for breathing effort and oxygen saturation (pulse oximetry) during recovery. Although significant quantities of fluid remain in the airways after BAL, most of the volume is absorbed rapidly. Residual amounts of fluid, however, can be retained for 24 to 48 hours after the procedure. During this time, some patients will manifest cough. Crackles may be auscultated.

Additional Reading
Hawkins EC: Bronchoalveolar lavage. In King LG, editor: Textbook of respiratory disease in dogs and cats, St Louis, 2004, Elsevier.
Hawkins EC, DeNicola DB, Plier ML: Cytological analysis of bronchoalveolar lavage fluid in the diagnosis of spontaneous respiratory tract disease in dogs, J Vet Intern Med 9:386–392, 1995.

Fine-Needle Aspiration of Lung
Percutaneous aspiration needle biopsy can be helpful in establishing a diagnosis in conditions such as (1) chronic inflammatory disease of the lung—for example, granulomatous lung disease caused by mycotic organisms; (2) chronic inflammatory disease; (3) metastases to the lung; and (4) primary lung tumors. The biopsy may provide enough diagnostic information to preclude performing an exploratory thoracotomy. Lung biopsy is contraindicated in animals with hemorrhagic disease or thoracic disease that produces forceful breathing and coughing.

Patient Preparation
Clip and surgically prepare the biopsy site. Infiltrate the skin, subcutaneous tissue, muscle, and parietal pleura with 1% to 2% lidocaine. In patients with diffuse parenchymal lung disease, taking biopsy material from the diaphragmatic lobes is recommended. The dorsal portions of the seventh to ninth intercostal spaces are preferred for percutaneous biopsies. In diffuse lesions, take biopsy material from the right or left thorax.

Caution: Understanding of the risks associated with performing fine-needle aspiration of the lung is important, and these risks must be clearly communicated to owners whose pets undergo this procedure. Lung aspirates will yield only cells, fluid, and trace amounts
of tissue, yet there is a significant risk of inducing pneumothorax with the procedure, even when performed without difficulty or complications.

**Technique**

For the procedure, a 22- to 25-gauge disposable needle (such as a 1-inch spinal needle) with stylet is preferred. Leave the stylet within the needle until the lung has been penetrated. Then quickly remove the stylet and immediately attach a sterile 6- to 12-mL syringe. The amount of air that might enter the lung between the time the stylet is removed and the syringe is attached is negligible. Holding the syringe carefully and steadily against the patient's thorax, establish negative pressure in the same manner as when obtaining an aspirate from a lymph node. As much as the patient will permit, attempt three to four aspirations without withdrawing the needle.

Alternatively, insert a conventional 25-gauge needle attached to a 6-mL syringe subcutaneously over the area of interest. Then establish significant negative pressure while the tip of the needle is still positioned in the subcutaneous tissues outside the parietal pleura. While maintaining the same amount of negative pressure in the syringe, direct the needle into the lung, leave it in place for 1 to 2 seconds, and withdraw it completely. Apply any material collected directly to glass slide. This procedure is best conducted in patients that are awake. Attempting the procedure in anesthetized dogs or cats could result in an unsuccessful aspiration, or, if the lungs were under positive pressure (ventilation or bagging), the risk of causing pneumothorax could be increased. Other reported complications include hemothorax (always exciting), lung laceration caused by patient movement during the procedure, pulmonary hemorrhage, and hemothysis. Contraindications to fine-needle aspiration include patients with a known bleeding diathesis and coagulopathy, thrombocytopenia, uncontrolled coughing, pulmonary hypertension, pulmonary cysts, and bullous emphysema.

Ultrasound-guided techniques for fine-needle aspiration or biopsy of the lung recently have been described and generally are associated with fewer procedural complications. However, additional training and experience, in addition to having access to the proper size and type of ultrasound probe, are critical.

**Additional Reading**

Cole SG: Fine needle aspirates. In King LG, editor: *Textbook of respiratory disease in dogs and cats*, St Louis, 2004, Elsevier.

**Nebulization and Aerosol Therapy**

Inhalation therapy can be defined as nebulization (humidification of the inspired air) and aerosol therapy (the process whereby drugs are vaporized in a solution and delivered directly into the respiratory tract). In companion animals, inhalation therapy is most useful for humidifying air in the respiratory tract and moistening the mucous membranes (nebulization). Sustained inspiration of dry air or gases causes irritation to the respiratory epithelium, which in turn results in swelling, bronchial gland hypertrophy, goblet cell proliferation, and loss of ciliary epithelium over time. Respiratory secretions become thick and tenacious, and efficient bronchial drainage is impaired.

The objectives of inhalation therapy include the following:

1. Humidification of bronchial mucous membranes
2. Deposition of miniscule amounts of potent drugs in smaller airways to achieve optimal topical therapeutic effects with minimal systemic side effects (e.g., bronchodilators)
3. Deposition of moderate amounts of potent agents or agents that are effective only topically (e.g., antibiotics and mucolytics)
4. Deposition of relatively large quantities of bland substances that promote bronchial drainage with minimal irritation (e.g., saline, propylene glycol, glycerin, and detergents)
Nebulization is used (1) in combination with oxygen therapy; (2) in tracheostomy care; (3) in acute respiratory diseases such as tracheobronchitis, bronchiolitis, upper respiratory disease of cats, pneumonia, and postoperative atelectasis and pneumonia; and (4) in chronic respiratory diseases such as chronic bronchitis, bronchopneumonia, collapsed trachea with secondary tracheobronchitis, emphysema, and bronchiectasis.

Aerosol therapy, however, is a limited-use therapeutic technique used in dogs and cats to administer antimicrobials, bronchodilators (aminophylline, 100 mg), or corticosteroids. The advantage of doing so is to achieve relatively high levels of drug in the respiratory tract in patients with defined lower respiratory tract disease. In addition, administration of potentially toxic antimicrobials (aminoglycosides) by this route has been shown to be associated with minimal or insignificant uptake into the general circulation, thereby minimizing (or eliminating) any risk of renal toxicity.

**Drug Delivery by Aerosolization** *(Figure 4-55)*

Drugs that can be applied by jet nebulizer *(Figures 4-56 and 4-57)* include the following:

1. **Bronchodilators:** Always use bronchodilators when administering drugs that may be irritating and constricting, such as isethionate hydrochloride 1% and phenylephrine 0.25%, 0.5 to 1.0 mL in 2 to 3 mL of saline three or four times daily.

2. **Antibiotics:** Antibiotics are poorly absorbed from the respiratory mucosa. Systemic administration of most antibiotics produces adequate pulmonary concentration for antibacterial effect. For *Bordetella* species that are located at the tips of bronchial cilia, topical contact via nebulization may be useful. Antibiotics that have been used successfully and safely include kanamycin (250 mg in 5 mL saline twice daily); gentamicin (50 mg in 5 mL saline twice daily); and polymyxin B (333,000 international units in 5 mL saline twice daily).

3. **Bland solutions:** Use these in large volume for prolonged mist effect: 0.9% sterile saline (5 to 200 mL as needed); glycerin (5% in saline); and propylene glycol (10% to 20% solution in saline).

4. **Detergents and mucolytics:** These compounds are irritating and currently are not recommended by most authors.

5. **Antifoaming agents:** Administer ethyl alcohol (70% solution, 5 to 10 mL twice daily).

*Figure 4-55:* Disposable jet nebulizer used to administer humidified air and/or medication directly into the respiratory tract.
Additional Reading
Boothe DM: Drugs affecting the respiratory system. In King LG, editor: Textbook of respiratory disease in dogs and cats, St Louis, 2004, Elsevier.
Tseng LW, Drobatz KJ: Oxygen supplementation and humidification. In King LG, editor: Textbook of respiratory disease in dogs and cats, St Louis, 2004, Elsevier.

URINARY TRACT PROCEDURES

UROHYDROPROPSLION
Removal of uroliths from dogs and cats is a commonly performed, yet critically important, clinical procedure. Several techniques are available for removal of calculi and obstructive concretions in male and female animals. Cystotomy is performed routinely...
to remove calculi from the lumen of the bladder but, especially in male dogs, may not be an effective approach for removing obstructing urethral calculi. Advanced and expensive techniques recently have been described: laparoscopic-assisted cystotomy, use of the Ellik evacuator, use of stone “baskets” (through a cystoscope), and lithotripsy are examples. However, for removal of uroliths from dogs and cats with partial or complete urinary obstruction, urohydropulsion is among the more effective yet inexpensive techniques available.

Urohydropulsion is a therapeutic procedure for removal of foreign material, namely, uroliths, from the bladder and/or urethra of dogs. Two techniques are described: voiding urohydropulsion and retrograde urohydropulsion. Both procedures have advantages and disadvantages.

**Voiding Urohydropulsion**

The objective of voiding urohydropulsion is to induce forceful voiding of urine by manually compressing the bladder to facilitate removal of cystic uroliths in female dogs.

*Caution:* Do not perform this procedure until it can be confirmed by catheterization or cystoscopy that the urethra is patent.

With the bladder filled with urine or saline (via catheterization), lift the patient (preferably sedated or anesthetized, although this procedure can be done in the awake patient) into a position such that the tail and perineum are ventral and the head is upright. The spine should be approximately perpendicular to the working surface. Using one or both hands, gradually increase pressure on the bladder to induce and maintain a forceful stream of urine. Objectively, small uroliths will be extruded. If the procedure is only partially successful, it can be repeated as necessary. Obviously, voiding urohydropulsion has limitations and cannot be used in male dogs or in dogs with urethral obstructions or strictures.

**Retrograde Urohydropulsion**

This procedure is indicated for male dogs and cats with partial or complete urethral obstruction caused by uroliths or accumulations of “sand.” This procedure should be performed in the anesthetized patient.

*Note:* There are discrepancies in the literature regarding whether to empty the urinary bladder of urine before performing this procedure. Because patients with urethral obstructions may have a significant volume of urine in the bladder at the time of presentation, some authors recommend performing cystocentesis to relieve the internal pressure before attempting urohydropulsion. However, in patients that have had a profoundly distended bladder for several hours (even days), penetrating the urinary bladder with a needle presents significant risk of rupturing a fragile bladder. The next step, of course, is abdominal surgery. I recommend avoiding cystocentesis whenever possible. The volume of saline required to flush uroliths into the bladder is inconsequential considering the total volume already present.

With the patient positioned in lateral recumbency, retract the prepuce and expose the penis as for conventional bladder catheterization technique. Use sterile technique to pass an appropriately sized flexible catheter, which is advanced to the point of obstruction. Attach a catheter-tipped 60-mL syringe filled with warmed (my preference) sterile saline and a water-soluble lubricant mixture (approximately two parts saline to one part lubricant) to the urinary catheter. An assistant places a gloved (always preferred) finger into the rectum to identify and occlude the lumen of the pelvic urethra at the level of the pubis. Subsequently, infuse saline forcefully into the catheter to dilate the urethra proximal to the obstructing urolith. At that point, release the digital pressure on the proximal urethra while the solution continues to be infused through the catheter. Objectively, the pressure within the urethra forces small stones retrograde into the urinary bladder, thereby relieving the obstruction.
Additional Reading

Adams LG, Syme HM: Canine lower urinary tract diseases. In Ettinger SJ, Feldman EC, editors: *Textbook of veterinary internal medicine*, ed 6, St Louis, 2005, Elsevier.

Osborne CA, Finco DR: *Canine and feline nephrology and urology*, Baltimore, 1995, Williams & Wilkins.