Chapter 18

Airway Management and Ventilation

Sandee M. Hartsfield

Introduction

Safe anesthesia includes establishment of a patient airway with assurance of adequate ventilation and oxygenation. If spontaneous ventilation is insufficient, the anesthetist should provide supplemental oxygen during the preanesthetic, induction, maintenance, and recovery phases of anesthesia.

Endotracheal Intubation

Indications

Indications for endotracheal intubation include maintenance of a patent airway, protection of the airway from foreign material, application of positive-pressure ventilation, application of tracheal or bronchial suction, administration of oxygen, and delivery of inhalant anesthetics. Placement of an endotracheal tube also reduces anatomical dead space if the tube is of the correct size and correctly positioned. For maintenance of inhalant anesthesia, an endotracheal tube should create a seal with the trachea to prevent leakage of anesthetic gases into the environment. An endotracheal tube is basic for endotracheal intubation, but ancillary equipment may be required in certain species. Intubation can be accomplished through the oral cavity, nasal passages, an external pharyngotomy, or tracheostomy.

Endotracheal Tubes

Murphy-type and Cole-type endotracheal tubes are commonly used in veterinary anesthesia. Uniquely, the Murphy tube has an opening, called a *Murphy eye* or *side hole*, in the wall opposite the bevel (Fig. 18.1); this hole allows gas flow, even if the end hole is occluded.[1] Characteristics of cuffed Murphy endotracheal tubes designed primarily for human patients are diagrammed in Fig. 18.2; such tubes are used in most veterinary patients for which appropriate sizes are available.

Cole tubes are uncuffed and are characterized by a shoulder near the distal end (Figs. 18.3 and 18.4); the diameter of the patient end of the Cole tube is smaller than the remainder of the tube.[1] Only the smaller portion of the tube should fit into the larynx and trachea. Although fitting the sloping shoulder of the Cole tube against the arytenoid cartilage creates a seal,[2] Dorsch and Dorsch indicate that, to avoid pressure against the laryngeal cartilages and to prevent laryngeal dilation, the shoulder should not contact the larynx.[1] The diameter of the tube should be such that the laryngotracheal part of the tube creates a seal, which guards against egress of gas and aspiration of foreign material. An effective seal can be established in veterinary patients of various sizes.[2,3]

Endotracheal tubes are made of polyvinyl chloride (PVC), rubber, silicone, and occasionally other plastic or rubberized materials. The most common tubes for human use are made of PVC,[1] many of which are used in small animals. Some endotracheal tubes, designed specifically for veterinary patients, are made of silicone rubber (Fig. 18.5). In general, endotracheal tubes should be clear so that they can be inspected for cleanliness or obstructions before each use. Red rubber tubes have been advertised for veterinary patients; such tubes are opaque, prone to cracking, and difficult to clean and disinfect.

Cuffed endotracheal tubes (Fig. 18.6) designed for human patients consist of a connector to fit the breathing system (15 mm

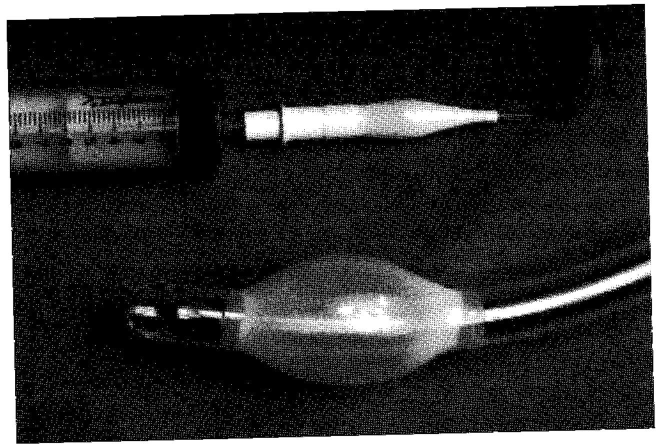


Fig. 18.1. A Murphy endotracheal tube characterized by a side hole (Murphy eye) opposite the bevel at the distal end of the tube. The inflatable cuff, pilot balloon, self-sealing inflation valve, and syringe for inflation of the cuff are shown. The parts and characteristics of a Murphy tube are diagramed and labeled in Fig. 18.2.

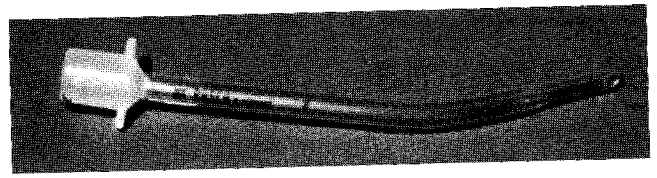


Fig. 18.3. A 10-French Cole endotracheal tube appropriate for small veterinary patients. Note the smaller diameter of the laryngotracheal portion of the tube (distal end of the tube, *right* side of the photograph).

OD [outer diameter]), the tube itself, and a cuff system (inflating valve, inflating tube, and pilot balloon). Labels on these tubes may include the manufacturer's name, internal and external diameters in millimeters, markings in centimeters indicating the length of the tube from the patient (distal) end, and IT, which indicates that the tube has been implantation tested. In addition, tubes labeled with either F29 or Z79 indicate that the tube material has been tested for tissue toxicity.[1,4] The terms *oral* and/or *nasal* may appear beside the tube's size for internal and external diameters, respectively. Some endotracheal tubes have the size in French units (French size = external diameter in millimeters times pi), which indicates the outside diameter of the tube. Radiopaque markers are embedded in some endotracheal tubes.

Inflation of the cuff of an endotracheal tube applies pressure to the tracheal mucosa. The perfusion pressure of the tracheal mucosa ranges from 25 to 35 mm Hg. A cuff pressure on the tracheal wall of 20 to 25 mm Hg will usually not interfere with tracheal mucosal blood flow.[5] Greater pressures in the cuff can lead to ischemic injury, mucosal damage, and ultimately tracheal strictures in serious cases. Therefore, the design of contemporary human endotracheal tubes includes a high-volume, low-pressure cuff that creates a good seal between the tracheal mucosa and the cuff wall when the cuff is properly inflated. The intent of the high-volume nonelastic cuff is to distribute the low-pressure seal over a relatively large area of the tracheal mucosa.[4] A cuff should be inflated with the smallest amount of air that will provide effective protection of the airway. A general recommendation is that pressure on the lateral wall of the trachea exerted by the cuff be maintained between 25 and 34 cm H_2O.[1] Generally, a leak should occur around the cuff when pressure equal to approximately 25 cm H_2O is applied to the airway.

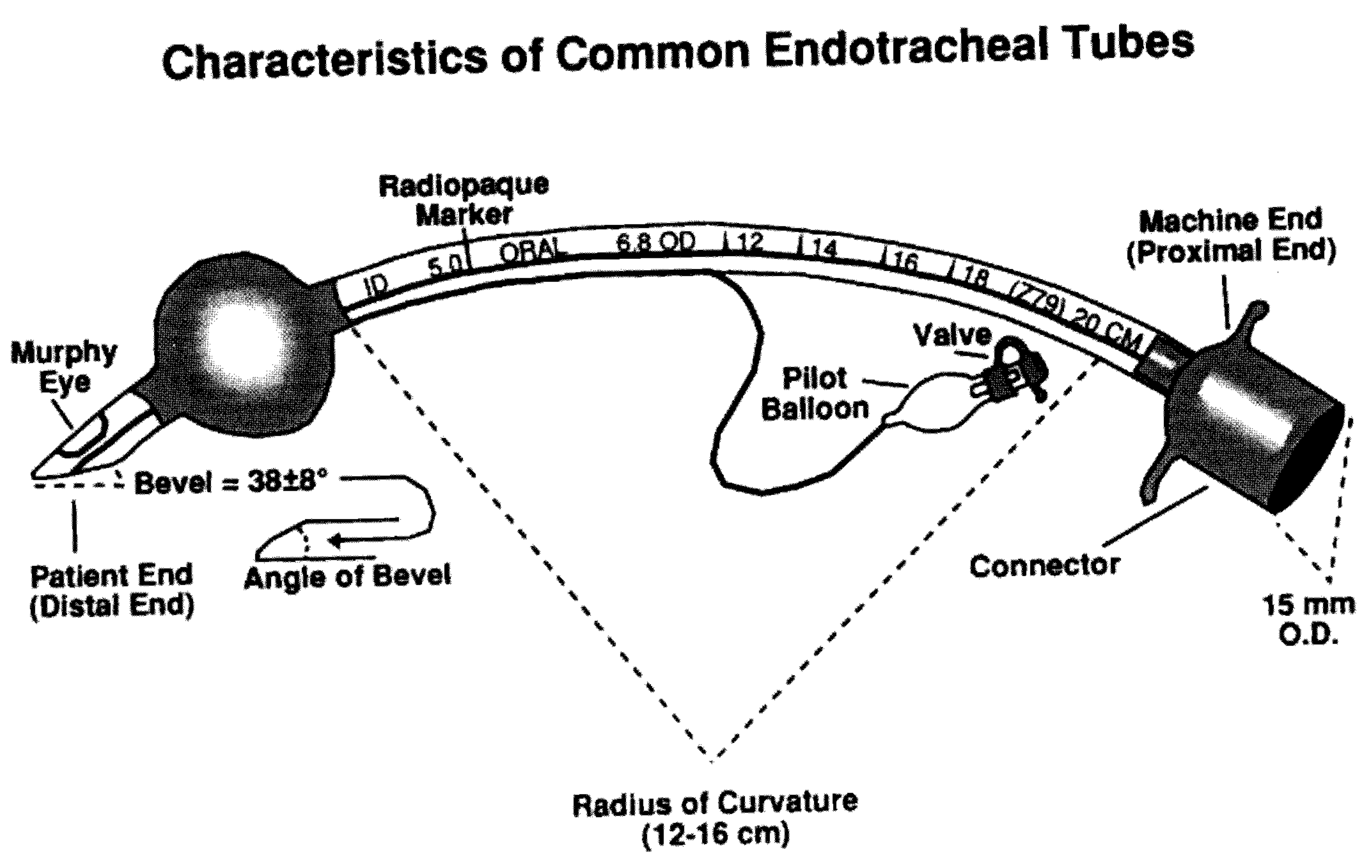


Fig. 18.2. Diagram illustrating the parts and desirable characteristics (e.g., radius of curvature and angle of the bevel) of a Murphy endotracheal tube. OD, outer diameter. From Dorsch and Dorsch.[1]

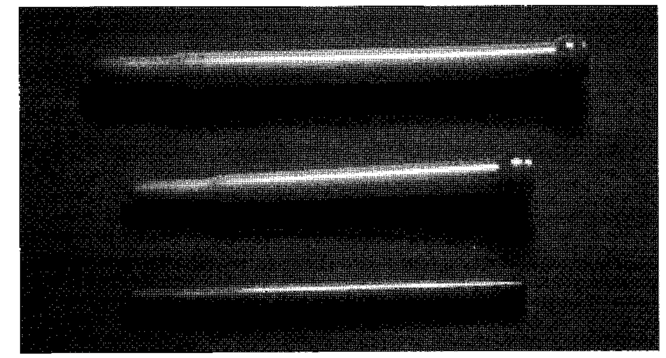


Fig. 18.4. Three sizes of Cole endotracheal tubes appropriate for large veterinary patients. Note the smaller diameter of the laryngotracheal portion of each tube (distal ends of the tubes, *left* side of the photograph). The proximal ends of the top two tubes are designed to fit the outside diameter of the Y piece of a circle breathing system for large animals.

Fig. 18.6. A Murphy endotracheal tube designed for human patients, but commonly used for small animals. Numbers and markings indicate the internal (5.0 mm) and external (8.0 mm) diameters, the length (13, 15, 17, 19, 21, and 23 cm) of the tube from the patient end, the manufacturer (Sheridan), and tissue toxicity testing (Z79). The internal diameter (5.0 mm) and manufacturer are also shown on the pilot balloon.

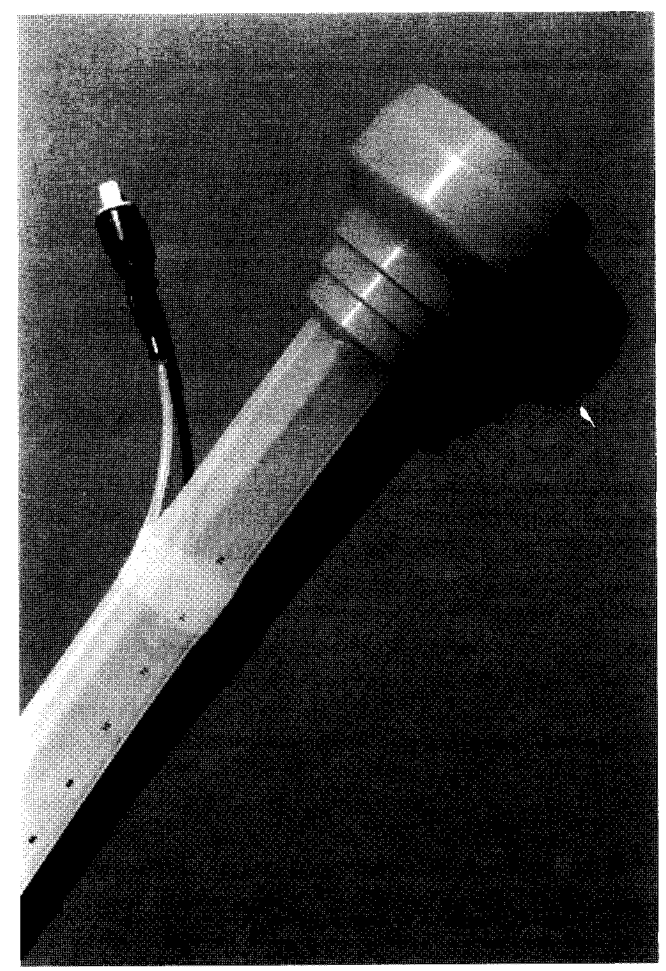


Fig. 18.5. Silicone rubber endotracheal tubes designed for veterinary use. The proximal end of the top tube has been fitted with a connector that will conform to the outside diameter of a Y piece of a circle breathing system for large animals.

Armored or reinforced endotracheal tubes (Fig. 18.7) are specially designed with helical wire or plastic implanted within the wall of the tube to prevent kinking of the tube and obstruction of the airway when the patient's head and neck are flexed. Such tubes are useful for ophthalmic surgery, cervical spinal taps, myelograms, oral surgery, and head and neck surgery. Armored tubes have thicker walls than standard tubes, causing them to have smaller internal diameters than standard tubes of equivalent external size.[4] Therefore, resistance to gas flow is increased, and reinforced tubes should not be used unnecessarily. Typically, these tubes are very flexible and more difficult to insert than standard PVC tubes. A stylet or guide tube will facilitate insertion of an armored tube into the larynx, but a stiff stylet should not extend past the distal end of the endotracheal tube.

Endotracheal intubation through a tracheostomy is sometimes necessary to provide a patent airway. Cuffed tracheostomy tubes with 15-mm-OD (outer diameter) connectors (Fig. 18.8) are available for use in human patients. However, standard endotracheal tubes for both large and small animals may be placed via

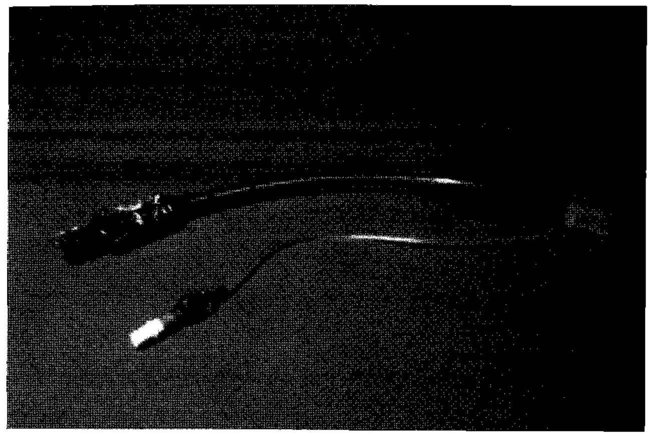


Fig. 18.7. An armored endotracheal tube with a spiral wire embedded in the wall of the tube: the endotracheal tube connector, bite guard near the proximal end of the tube, inflatable cuff, pilot balloon, inflation line, and self-sealing inflation valve.

Fig. 18.9. A silicone rubber endotracheal tube placed through a tracheostomy site to facilitate inhalant anesthesia for oral and nasal surgery in a foal.

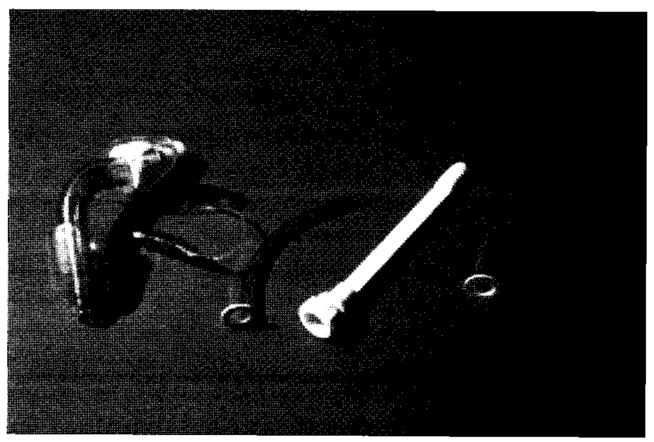


Fig. 18.8. A cuffed tracheostomy tube (*left*) designed for human patients, but applicable to veterinary patients. From *left to right* are the cuffed tracheostomy tube with inflation line and pilot balloon, a removable lumen for the tube, an obturator to facilitate insertion of the tube, and another removable lumen.

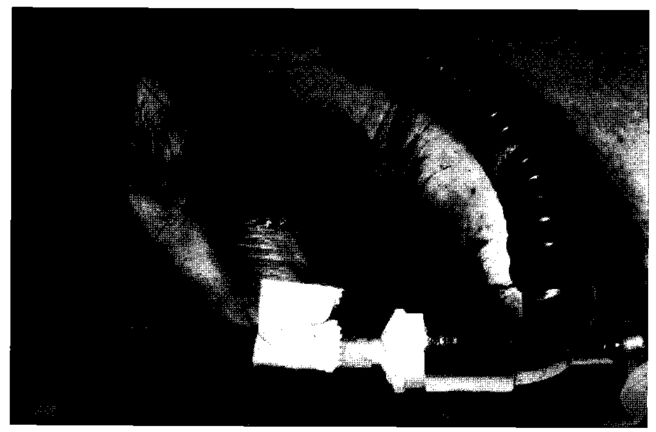


Fig. 18.10. An endotracheal tube placed by external pharyngotomy in a small dog to facilitate oropharyngeal surgery. From Hartsfield.[19]

tracheostomy to facilitate general anesthesia (Fig. 18.9). Endotracheal tubes are also recommended for intubation by external pharyngotomy (Fig. 18.10).[6]

Normally, endotracheal intubation is accomplished in anesthetized patients. Forced intubation in awake or lightly anesthetized patients should be avoided unless dictated by special circumstances. Traumatic intubation can produce laryngeal edema, laryngeal spasm, hemorrhage, and vagal stimulation leading to bradycardia and other arrhythmias. Direct application of a local anesthetic (e.g., lidocaine [Fig. 18.11]) to the larynx may prevent laryngeal spasms in susceptible animals (e.g., cats and swine). The local anesthetic can be sprayed into the larynx, applied with a cotton swab, or squirted from a syringe and hypodermic needle. When using a syringe, the hypodermic needle must be firmly attached to prevent its dislodgment and entrance into the larynx. The total dose of local anesthetic should not approach a toxic dose, based on the patient's species and body weight. In very small patients, spraying local anesthetic onto the larynx can easily exceed a toxic dose.

Endotracheal and tracheostomy tubes should be cleaned thoroughly after use. The tubes should be gently scrubbed with a soft brush, rinsed, dried, and sterilized or disinfected. If ethylene oxide is not available, chemical disinfectants can be used; glutaraldehyde has been recommended.[7] After disinfection, tubes should be rinsed thoroughly, according to recommendations for use of the disinfectant, and dried. If tubes are inadequately

Fig. 18.11. Lidocaine as a spray (*left*) or as a liquid (applied with a cotton swab or delivered by squirting it with a syringe and needle) can be applied topically to the larynx to facilitate endotracheal intubation in various species. With either method of delivery, the anesthetist should keep the total dose of lidocaine less than the toxic dose for the patient and species involved.

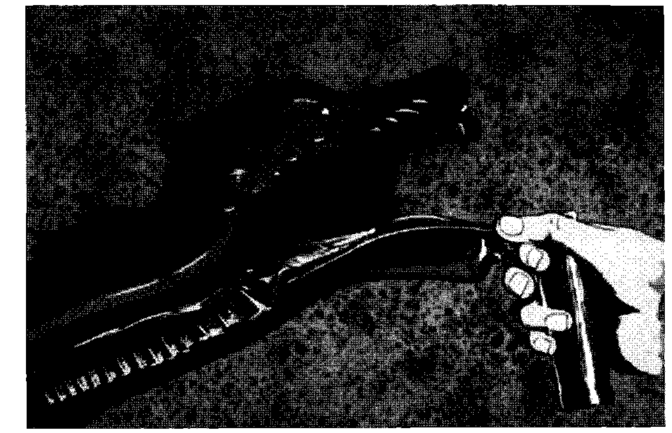


Fig. 18.12. Diagram of the correct positioning of a laryngoscope blade for maximum visualization of the larynx. Note that the dog's mouth is opened widely, its tongue is extended from its mouth maximally, and the tip of the laryngoscope blade is positioned at the base of the epiglottis.

rinsed, tissue reactions to the disinfectant can occur. Ethylene oxide sterilization should be performed according to the manufacturer's recommendations for the product and its sterilization equipment. Endotracheal tubes should be clean and dry before they are sterilized with ethylene oxide. Appropriate aeration time should be allowed between sterilizing a tube and its use in a patient: typically 2 days at 120°F (49°C) in an aeration chamber or 14 days if no aeration chamber is available. Failure to allow sufficient aeration time can cause serious respiratory complications.[8] Most endotracheal or tracheostomy tubes should not be autoclaved; however, silicone rubber tubes can be steam autoclaved.[7,9]

Laryngoscopy is required for endotracheal intubation of some species. Often, the laryngoscope's light source is the main benefit of laryngoscopy, but the blade can be used to manipulate the tongue, soft palate, and epiglottis to view the glottis (Fig. 18.12). Useful blades include the Miller, the McIntosh, and the Bizarri-Guiffrida (Figs. 18.13 and 18.14), and other blades are available. Different lengths of blades are needed for various species. As examples, very short blades designed for human infants are useful in rabbits, and blades up to 205 mm designed for human adults are appropriate for large dogs. Specially designed, very long (350 to 450 mm) blades can be purchased for veterinary use and may be needed in llamas, cattle, swine, and other species.

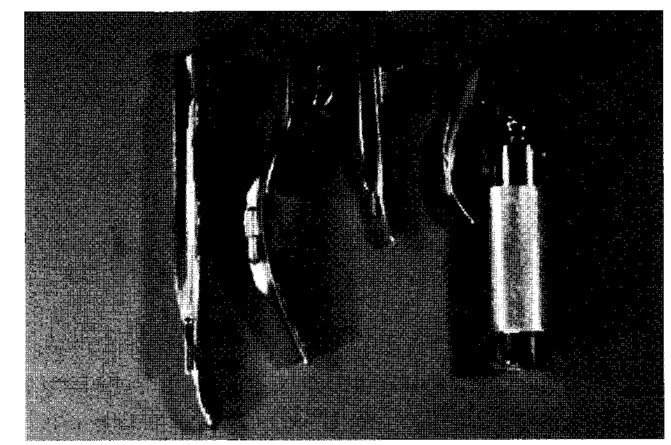


Fig. 18.13. From *left to right*, an adult Miller laryngoscope blade, an adult Bizarri-Guiffrida blade, a pediatric Miller blade, and a pediatric Bizarri-Guiffrida blade on a laryngoscope handle. The Bizarri-Guiffrida blades allow the maximum field of view without the interference of a flange.

Techniques of Endotracheal Intubation

Dogs

For most dogs, an endotracheal tube and adequate lighting are the only necessities for intubation of the trachea. However, the use of a laryngoscope, a stylet to stiffen the endotracheal tube, a guide tube (Fig. 18.15), sterile water-soluble lubricant, a mouth speculum, and local anesthetic may be desirable and even necessary under certain circumstances.

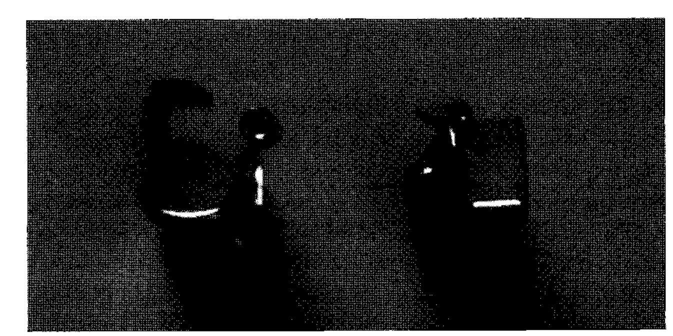


Fig. 18.14. End-on view of Miller (*left*) and Bizarri-Guiffrida (*right*) laryngoscope blades. The Bizarri-Guiffrida blade allows maximum space for passage of the endotracheal tube, and the Miller blade provides a flange to elevate redundant tissue (e.g., soft palate).

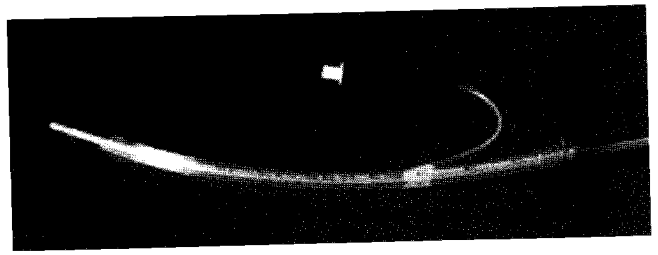


Fig. 18.15. Photograph of a silicone rubber endotracheal tube with a 10-French canine polyethylene urinary catheter preplaced for use as a guide tube. The guide tube will pass easily through the larynx and into the cranial part of the trachea to facilitate passage of the endotracheal tube.

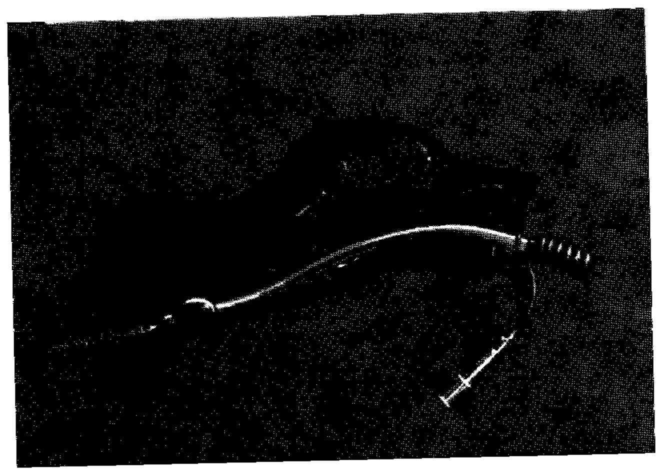


Fig. 18.16. Diagram of the correct placement of an endotracheal tube in a dog. Note that the connector is located near the incisor teeth to minimize mechanical dead space and that the cuffed end of the tube is in the cervical trachea near the thoracic inlet.

Sizes of endotracheal tubes for canine patients range from 1.5 mm to approximately 15 mm ID (internal diameter).[2] It is difficult, if not impossible, to find cuffed tubes smaller than 3.0 mm ID. There are breed differences that preclude generalizations about the choice of tube diameter based on a patient's body weight or some other arbitrary guide. For example, a 25-kg English bulldog usually accepts only about a 7.5-mm-ID endotracheal tube, but a 25-kg mixed-breed dog may easily accept a 10-mm tube. Most tubes designed for human patients are too long for dogs and should be cut at the proximal end to fit the patient; the connector should be positioned at the level of the dog's incisors, and the distal end should be located in the trachea near the thoracic inlet (Fig. 18.16).

After induction of anesthesia, the dog is positioned in sternal recumbency for intubation. An assistant holds the dog's head with one hand, placing the finger and thumb behind the maxillary canine teeth and pulling the dog's lips upward to create the best field of view. With the other hand, the assistant opens the dog's mouth widely and extends its tongue. The assistant should not put pressure under the dog's neck because the view of the larynx will be obstructed by the soft palate. With a good light source, most dogs can be intubated without a laryngoscope. If an assistant is unavailable, an oral speculum will keep the dog's mouth open during intubation. In small patients, dogs with oral or pharyngeal lesions, and brachycephalic dogs, a laryngoscope facilitates intubation and should always be available if difficulty should arise. The endotracheal tube should be secured to prevent its dislocation during anesthesia. Using a piece of rolled gauze, the tube can be tied to the maxilla (Fig. 18.17), the mandible, or behind the head, depending on the breed, the type of surgery, the presence and condition of the canine teeth, and the anesthetist's preference.

Fig. 18.17. An endotracheal tube secured to a dog's maxilla with a piece of rolled gauze. Note that the gauze is tied tightly around the tube without constricting the lumen, that the connector is at the level of the incisor teeth, and that the gauze is positioned immediately caudal to the maxillary canine teeth and tied in a bow.

Extubation should be done when the dog's oral and pharyngeal reflexes have returned. The tube should be pulled directly between the upper and lower incisor teeth. If a tube is allowed to deviate laterally, the dog may shear the tube. This damages tubes and creates the potential for aspiration or ingestion of a part of the tube.

Cats

The primary equipment required for feline intubation are an endotracheal tube and a light source. However, a laryngoscope, a stylet to stiffen the endotracheal tube, a guide tube (canine polyethylene urinary catheter), sterile water-soluble lubricant, a mouth speculum, and local anesthetic may be useful. If a wire stylet is used to stiffen the tube, the stylet should not extend past the distal end of the tube to avoid injury to the trachea.[2] Inadequate depth of anesthesia is probably the most common reason for difficult intubation.

Sizes of endotracheal tubes for domestic cats range from 1.5 mm to approximately 5.5 mm ID; most adult cats readily accept 4.0- to 4.5-mm-ID tubes, a range that provides optimal internal diameter with minimal difficulty in intubation. It is difficult to find cuffed tubes smaller than 3.0 mm ID, and such sizes may be needed for small kittens. One option is to use small Cole tubes. Since most endotracheal tubes designed for human patients are too long for cats, the tube should be cut at the proximal end to fit the patient. The proximal end of the tube should be positioned at the level of the cat's incisors, and the distal end should be located in the trachea near the thoracic inlet.

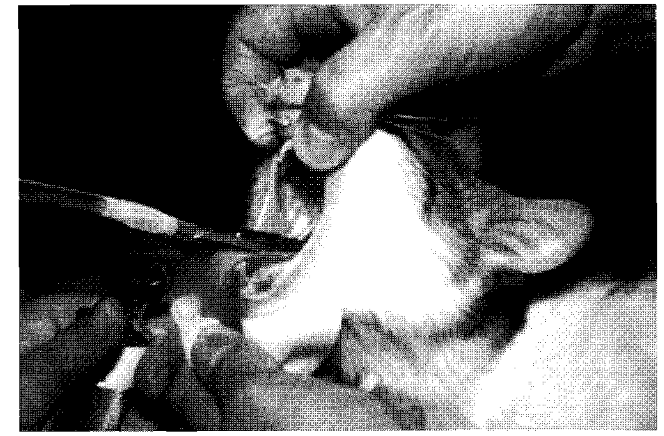


Fig. 18.18. Illustration of an excellent method of positioning a cat for endotracheal intubation. Note the secure grip on the maxilla with the index finger and thumb caudal to the canine teeth. The tongue is extended, maximizing the field of view.

Fig. 18.19. View of a cat's glottis using the restraint and positioning depicted in Fig. 18.18. The laryngoscope blade is placed on the tongue with the tip just ventral to the epiglottis.

After induction of anesthesia, the cat should be positioned in sternal recumbency. Although not necessary in every case, local anesthetic (0.5% lidocaine) may be applied to the larynx to desensitize the arytenoid cartilage and epiglottis to help prevent laryngospasm during intubation. An assistant holds the head with one hand, placing a finger and thumb behind the cat's maxillary canine teeth and pulling the lips upward to create the best field of view (Figs. 18.18 and 18.19). With the other hand, the assistant extends the cat's tongue. If the tongue is not protruding from the mouth, the laryngoscope blade can be used to manipulate the tongue so that the assistant can grasp it. Neither the anesthetist nor the assistant should put their fingers into a lightly anesthetized cat's mouth. The assistant should not put pressure under the cat's neck because the view of the larynx may be obstructed by the soft palate. As in dogs, with a good light source, most cats can be intubated without the aid of a laryngoscope. However, a laryngoscope is often helpful. The blade should not touch the arytenoid cartilage or the epiglottis (Fig. 18.18) because such stimulation may cause active closure of the glottis. A laryngoscope should always be available for a difficult intubation (e.g., oral or pharyngeal lesions). If an assistant is unavailable, an oral speculum will keep the cat's mouth open while intubation is accomplished. The routine use of a guide tube (5- to 8-French canine urinary catheter) that extends past the cuffed end of the endotracheal tube (Fig. 18.18) for 2 or 3 cm often makes feline intubation easier. As the endotracheal tube is advanced toward the glottis, rotating it from 0° to 90° or greater will facilitate its passage. Rolled gauze can be used to secure the tube behind the cat's head with a simple bow knot. In cats, the tube should be tied for rapid removal at recovery.

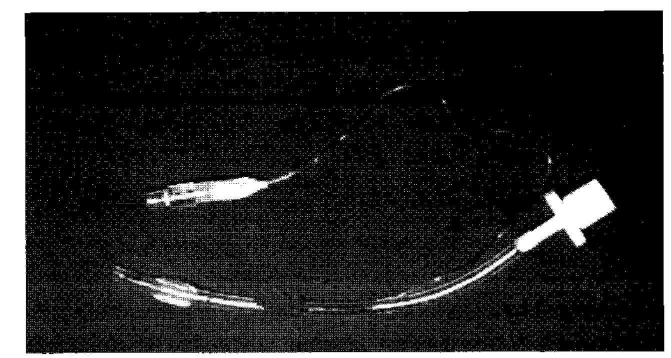


Fig. 18.20. A endotracheal tube (4-mm internal diameter) sheared during extubation of a cat at the time of recovery from anesthesia. Aspiration or ingestion of the smaller piece is possible.

Extubation should be done when the cat's oral and pharyngeal reflexes have returned. The tube should be pulled directly between the upper and lower incisor teeth. If a tube is allowed to move laterally, the cat may shear the tube (Fig. 18.20), which creates the potential for aspiration or ingestion of part of the tube.

Horses

Blind passage of the endotracheal tube in horses can be aided by a mouth speculum. For routine intubation, PVC connectors (10 cm long, variable diameters) for PVC pipe make economic, effective specula that can be placed between the horse's upper and lower incisors to protect the tube. The connectors can be wrapped with adhesive tape to increase friction between the teeth and the speculum.[7] Other supplies that may be useful for equine intubation under certain conditions include a guide tube (equine stomach tube), sterile water-soluble lubricant, local anesthetic, and a fiber-optic endoscope.

To assure an appropriate range of sizes of endotracheal tubes for equine patients (miniature horses to draft horses), tubes as small as 7 mm ID and as large as 30 mm ID should be available. Larger sizes (e.g., 35 mm ID) have been recommended for large thoroughbred and draft horses.[10] Tube size varies with the size of the patient and with the location of the tube (oral versus nasal intubation). Modern 26-mm-ID silicone rubber cuffed endotracheal tubes are appropriate for a high percentage of adult horses, with 30-mm tubes indicated for very large horses. In general, a tube that is passed nasally should be about two sizes smaller than a tube that is passed orally.[9]

In preparation for oropharyngeal intubation, the horse's mouth should be flushed with water to remove any debris that may be retained in the oropharynx, including the cheek pouches. Horses are positioned in lateral recumbency for intubation with a lubricated endotracheal tube. A sterile water-soluble lubricant should be used; lubricants containing local anesthetic are unnecessary and may irritate airway tissues.[7,11] The mouth speculum is placed between the upper and lower incisors, the head and neck are extended, and the endotracheal tube is advanced into the pharynx until the tip of the tube touches the larynx. In some patients, the tube enters the larynx without any interference. However, several attempts (a series of 10- to 15-cm advancements and retractions of the tube, with rotation of the tube from 0° to 90° or greater as it approaches the glottis) may be necessary for intubation, even in normal horses. Although the technique is somewhat of an art, intubation can be facilitated by maximally extending the horse's head and neck in a straight line with the its back (best done by an assistant), extending the tongue during intubation, and holding the endotracheal tube so that the proximal end is curved below the mandible during attempts at intubation. In general, an endotracheal tube of proper size can be passed into the larynx with little if any resistance once correct positioning and technique have been established.

For difficult intubation, an equine stomach tube (guide tube) may be passed into the larynx and trachea, over which the endotracheal tube can be manipulated through the larynx and into the trachea. In some instances (e.g., laryngeal or pharyngeal abnormalities), equine intubation may be successful only after visualizing the glottis with a fiber-optic endoscope; this allows adjustments in the position of the endotracheal tube or a guide tube as it approaches the glottis.

A properly positioned endotracheal tube is usually obvious to an experienced anesthetist because of the absence of resistance as the tube enters the larynx and trachea. Air flow into and out of the tube during spontaneous ventilation can be used to verify tube placement. Some veterinarians advocate compression of the thorax to create air flow from a properly placed tube, but this technique may not be foolproof. Finally, water may condense on the inner surface of the tube during exhalation if the tube is placed properly.

Swine

Endotracheal intubation of swine is relatively difficult for several reasons:[12] The distance from the tip of the snout to the larynx is comparatively long, the mouth does not open widely, the larynx is rather loosely attached, mobile, relatively small, and slopes ventrally, creating a sharp angle for passing an endotracheal tube (Fig. 18.21). In addition, laryngospasm is rather easily induced in lightly anesthetized pigs. An endotracheal tube, a laryngoscope, 2% lidocaine in a syringe, sterile water-soluble lubricant, and a guide tube should be available for intubation of swine.

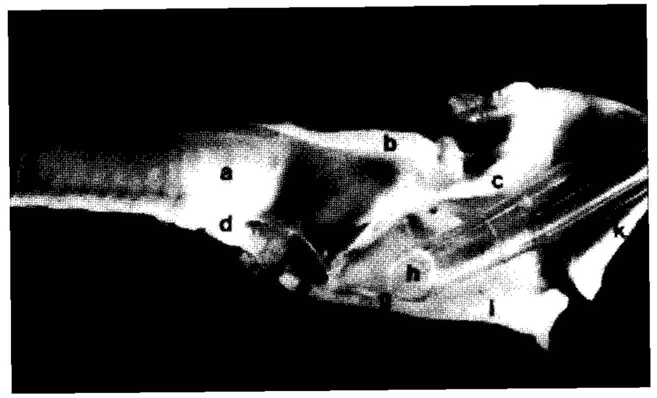


Fig. 18.21. Sagittal section of a pig's larynx. This illustrates the irregular course that an endotracheal tube must travel as it moves through the larynx and into the cranial trachea: *a*, tracheal opening; *b*, dorsal cricoid cartilage; *c*, arytenoids cartilage; *d*, ventral cricoid cartilage; *e* and *i*, thyroid cartilage; *f*, entrance to lateral laryngeal ventricle; *g*, posterior floor of the larynx; *h*, tip of endotracheal tube; *j*, middle laryngeal ventricle; and *k*, and epiglottis. Reproduced by permission of Dr. William Tranquilli.

Compared with other domestic species, swine have small laryngeal and tracheal diameters. Endotracheal tube sizes from 3 mm ID in piglets to 16 mm ID in larger swine may be needed. Mature sows and boars may accept even larger tubes. After induction of anesthesia, the pig is placed in sternal recumbency, an assistant holds the head with a small rope or piece of rolled gauze passed through the mouth, and the pig's tongue is extended. A mouth speculum can be employed, if necessary. Using the appropriate length of blade, the pig's larynx is visualized with the aid of a laryngoscope; this usually requires some manipulation to position the blade for a good view of the glottis, especially in large swine with a narrow pharynx and excessive tissue in the area of the soft palate. Lidocaine can be squirted onto the larynx for desensitization. The guide tube (usually two 10-French canine urinary catheters in tandem) is passed through the larynx and into the trachea; the guide tube should be manipulated through the larynx without excessive force, and best results are obtained by passing the guide tube along the dorsal aspect of the larynx to the midcervical trachea. Then, a well-lubricated endotracheal tube is directed over the guide tube, through the larynx, and into the trachea; firm, gentle advancement of the tube with a simultaneous twisting motion (0° to greater than 90°) is helpful. The tube should be positioned with the connector at the level of the tip of the snout, and the distal end should be near the thoracic inlet. The cuff should be inflated with the minimum amount of air that will create a seal. The tube can be secured to the snout with adhesive tape or behind the ears with rolled gauze.

Miniature pet pigs are intubated by using the same equipment (smaller sizes) and method just described, but gentle technique should be emphasized. Laryngospasm, laryngeal edema, and death have been associated with traumatic intubation in miniature pigs.[13]

Cattle

The primary implements for endotracheal intubation in adult cattle are endotracheal tube, a mouth speculum (e.g., Bayer dental wedge, Guenther mouth speculum, Weingart mouth speculum, or Drinkwater mouth gag), and an equine stomach tube (two to three times longer than the endotracheal tube) for use as a guide. For smaller cattle, a long laryngoscope blade (e.g., 14, 16, or 18 inches) may be necessary for passage of a guide tube. Sizes of endotracheal tubes ranging from 18 to 30 mm ID may be needed for adult cattle.

After induction of anesthesia, a mouth speculum is positioned to hold the mouth open; this helps to prevent damage to the endotracheal tube cuff and to the anesthetist's hand and arm during intubation. The cow's tongue is extended from the mouth as its head and neck are extended. The anesthetist passes one hand through the cow's mouth and palpates the epiglottis and glottis. The anesthetist passes the guide tube into the pharynx and then slides the tube through the glottis, assuring the tube's proper placement by palpation as it enters the larynx. The guide tube is advanced until its tip is in the midcervical trachea. After the anesthetist's arm is removed from the cow's mouth, the endotracheal tube is advanced over the guide tube and into the cow's larynx and trachea. Rotation of the tube from 0° to greater than 90° as the tube approaches the arytenoid cartilage will help to advance the endotracheal tube into the trachea. The cuff should be inflated immediately to decrease the likelihood of aspiration of regurgitated rumen contents. Should active or passive regurgitation of large quantities of ruminal content occur just before or simultaneously with endotracheal intubation, external pressure applied over the esophagus will halt the flow of ruminal contents. Alternatively, the endotracheal tube can be quickly passed into the esophagus and the cuff inflated, permitting the regurgitant to flow through the endotracheal tube beyond the pharynx and out of the mouth, preventing its aspiration into the lungs. A properly positioned endotracheal tube in a cow is shown in Fig. 18.22.

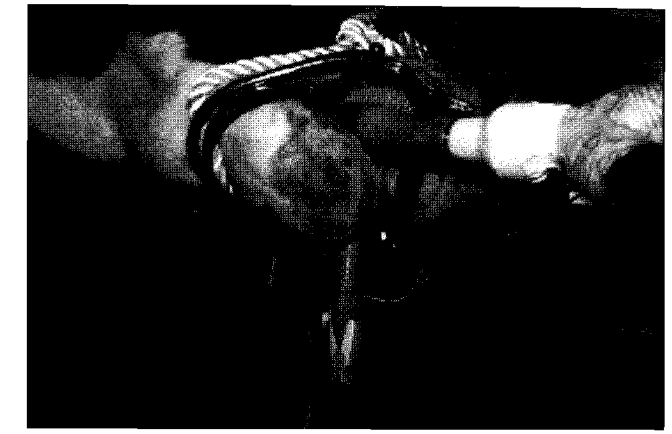


Fig. 18.22. A silicone rubber endotracheal tube (26-mm internal diameter) placed in a cow. The tube was passed with the Wiengart mouth speculum in place as shown.

Bovine intubation can be accomplished without a guide tube. The endotracheal tube is passed beside or under the anesthetist's arm and palpated as it enters the cow's larynx.[12] Alternately, the anesthetists can take the tube into the cow's mouth, cupping the distal end of the tube in the hand.[14] The disadvantage of either method is that the size of the endotracheal tube that can be readily passed is limited, especially if the anesthetist has a large arm.

Small Ruminants

Equipment required for endotracheal intubation in small ruminants includes an endotracheal tube, a laryngoscope, and a guide tube. Endotracheal intubation in small ruminants (sheep, goats, calves, cattle weighing less than about 250 kg, deer, and exotic ruminants) is best accomplished by direct visualization of the larynx with an illuminated laryngoscope. Sternal recumbency facilitates the procedure, but intubation can be achieved during lateral recumbency. After induction of anesthesia, an assistant holds the animal's head while the anesthetist extends its tongue. The anesthetist passes the laryngoscope blade over the base of the tongue to visualize the glottis, and then passes a guide tube (e.g., a small equine stomach tube in calves or a polyethylene catheter in sheep or goats) into the larynx and into the trachea to about the midcervical area. The laryngoscope is removed, and the endotracheal tube is passed over the guide tube and into the trachea. The cuff is inflated immediately to decrease the likelihood of aspiration of regurgitated rumen contents. The use of a metal rod has been advocated as a guide tube.[14] Excessive force with a metal guide tube increases the risk of damaging the larynx or trachea and is not recommended.

Nasotracheal Intubation

This is commonly used in foals for the administration of inhalant anesthetics during induction.[9] The technique can be also be used in calves and adult horses, and its use has been described in llamas.[15]

The characteristics of an ideal nasotracheal tube include a tube with minimal curvature and extra length (55 cm). The tube should be made of inert material (e.g., silicone rubber) and have relatively thin walls for maximum internal diameter. The tube should resist kinking. Low-volume, high-pressure cuffs may be less traumatic during placement, but high-volume, low-pressure cuffs may be best for longer periods of anesthesia. Tubes as small as 7 mm ID may be necessary for neonatal foals. In general, in any given patient a nasotracheal tube should be one to two sizes smaller than the appropriately sized orotracheal tube.[9] Once induction is completed using a nasotracheal tube, the nasotracheal tube can be removed and replaced with an appropriately sized orotracheal tube to decrease resistance to gas flow.

Nasotracheal intubation (Figs. 18.23 through 18.26) involves passage of a properly sized endotracheal tube through the nostril

Fig. 18.23. Restraint of an nontranquilized foal for nasotracheal intubation. A lubricated tube is directed through the ventral meatus.

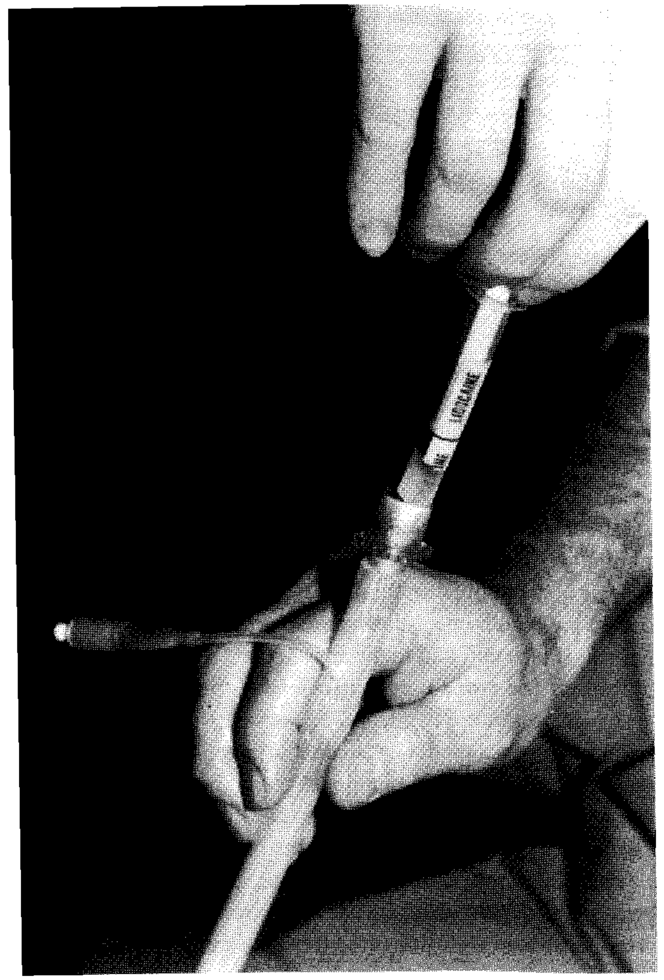


Fig. 18.24. Lidocaine (2%) being injected through the endotracheal tube, the distal end of which is near the foal's glottis. The lidocaine should desensitize the epiglottis and arytenoid cartilages to facilitate passage of the tube through the glottis.

(Fig. 18.23), ventral nasal meatus, and larynx and into the trachea. Lidocaine gel (10%) is a good lubricant for the tube and should be applied to the nostril and rostral portion of the nasal passage before advancing the tube in awake animals. A sterile water-soluble lubricant without lidocaine is appropriate for anesthetized patients. With the patient's head and neck extended, the tube is advanced into the pharynx and passed into the larynx on inspiration. Air moves freely through a correctly placed tube during spontaneous ventilation. Taping the tube to the muzzle is appropriate (Fig. 18.26).

For uncooperative foals and calves, sedation may facilitate nasotracheal intubation. Some awake patients cough and close the glottis in response to the tube contacting the larynx. With the nasotracheal tube positioned with the cuffed end near the larynx, 2% lidocaine solution can be flooded onto the larynx via the tube (Fig. 18.24). This desensitizes the larynx and eases intubation.

Extubation following nasotracheal intubation should be done carefully. After deflation of the cuff, the tube should be withdrawn slowly and deliberately, with the patient's head restrained to avoid any sudden, jerky motions. Rapid, rough extubation may cause unnecessary nasal hemorrhage.

Rabbits and Other Laboratory Animals

Intubation techniques for rabbits and other small laboratory animals have been described.[16] Most techniques for intubation of small laboratory animals include the use of a laryngoscope or a modified otoscope to expose the glottis, a catheter or stylet to serve as a guide tube, a small-diameter lubricated endotracheal tube, and lidocaine to desensitize the larynx before passing the endotracheal tube. In laboratory rabbits weighing about 3.0 kg, the use of 3.5-mm-ID, 14-cm-long endotracheal tubes are appropriate.[17]

The rabbit has been described as perhaps the most difficult animal to anesthetize.[18] Undoubtedly, problems with airway management influenced that opinion. However, the technique for endotracheal intubation in rabbits can be mastered with practice when using proper equipment. Guide-tube technique causes minimal trauma during intubation and allows selection of the largest suitable endotracheal tube. Endotracheal intubation in rabbits requires gentle manipulations. Rough technique invariably leads to

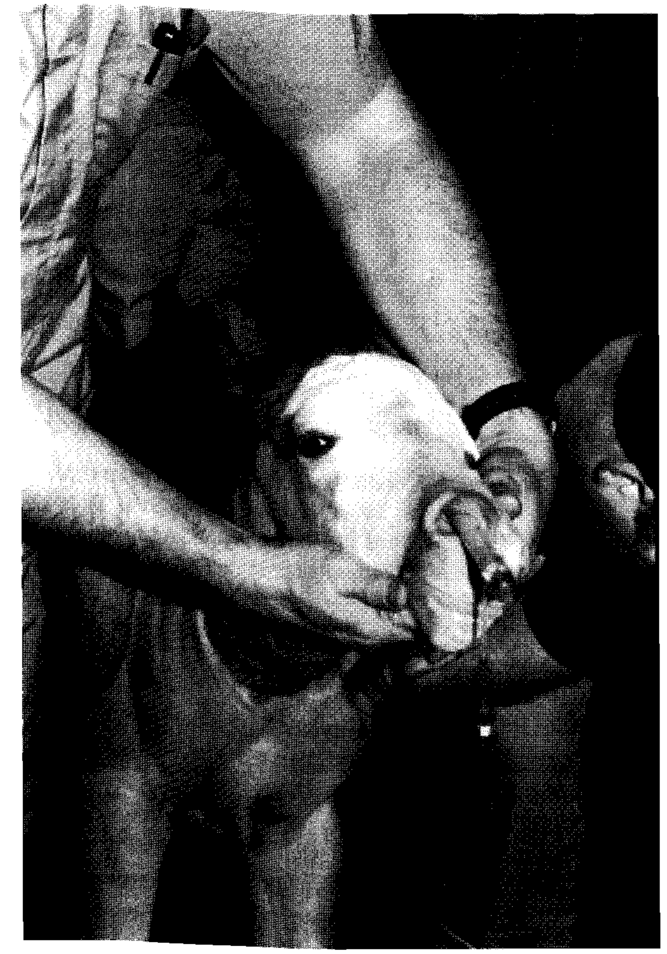


Fig. 18.25. A nasotracheal tube is positioned in the trachea and ready to be secured to the patient. The proximal end of the tube extends a few centimeters from the nostril to facilitate taping (see Fig. 18.26).

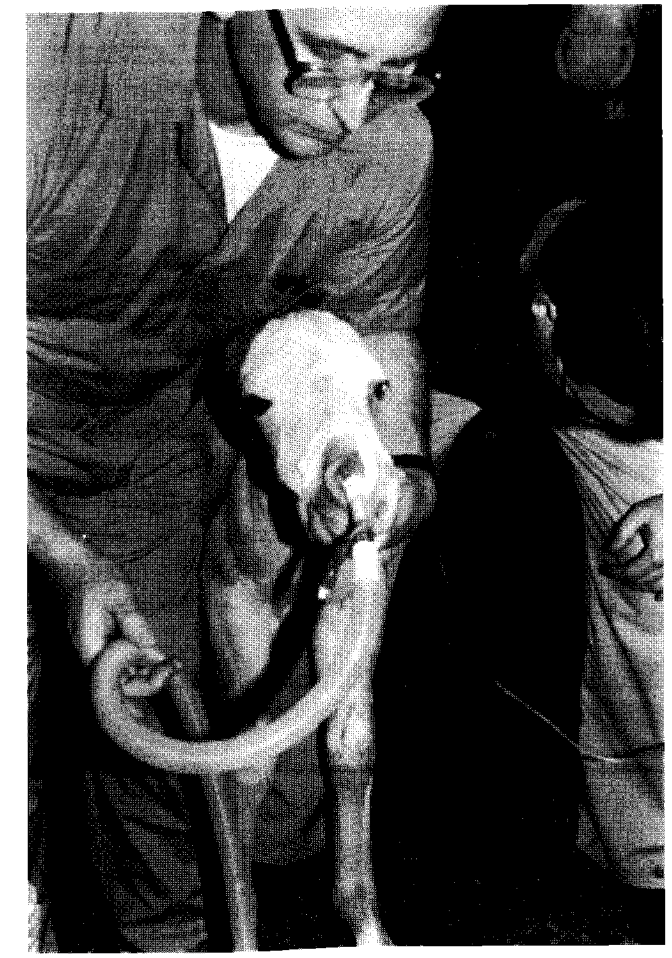


Fig. 18.26. A nasotracheal tube secured to a foal's muzzle. The cuff has been inflated, and an adult Bain breathing system has been connected to the endotracheal tube to facilitate induction of anesthesia with isoflurane in oxygen.

trauma to the tongue, pharynx, larynx, or trachea. Trauma with associated edema and hemorrhage can cause lethal complications.

Rabbits can be intubated when positioned in sternal recumbency with the head and neck extended and the fleshy tongue gently withdrawn from the mouth (Figs. 18.27 to 18.30). The rabbit's head can be held with a piece of rolled gauze placed caudal to the maxillary incisors. A size-0 Miller laryngoscope blade (75 mm long) is used to expose the glottis; the blade is carefully manipulated lateral to the maxillary incisors, into the mouth, and over the base of the tongue to expose the soft palate, epiglottis, and glottis. All are very fine, but distinct, anatomical structures. The epiglottis may be positioned behind the soft palate. The anesthetist should definitively identify the glottis before proceeding. Then, a guide tube (a 5- to 8-French canine urinary catheter) can be passed through the larynx and into the midcervical trachea, about 2 cm past the glottis (Fig. 18.27). The guide tube should not be forced because the trachea is easily torn, which can eventually lead to subcutaneous emphysema, pneumothorax, pneumomediastinum, pneumoabdomen, or death. The endotracheal tube is passed over the guide tube, through the larynx, and

Fig. 18.27. Endotracheal intubation in a rabbit. The rabbit is positioned in sternal recumbency, the glottis has been exposed with a size-0 Miller blade, and the anesthetist is passing a 7-French canine urinary catheter to serve as a guide tube for intubation.

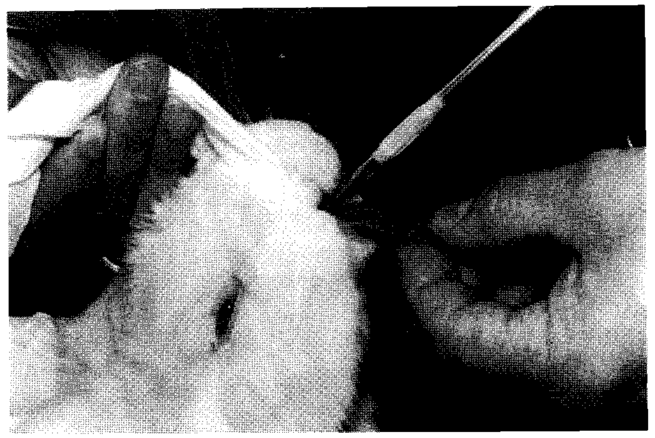


Fig. 18.28. Endotracheal intubation in a rabbit. The endotracheal tube is advanced over the guide tube and into the mouth (the distal end of the guide tube is located 2 cm caudal to the cricoid cartilage).

Fig. 18.29. Endotracheal intubation in a rabbit. The distal end of the endotracheal tube is located just rostral to the glottis, the tip of the guide tube is in the cervical trachea, and lidocaine is flushed through the lumen of the endotracheal tube to desensitize the larynx before advancement of the endotracheal tube.

into the trachea. If resistance to passing the endotracheal tube through the glottis is apparent, less than 0.5 mL of 2% lidocaine can be flushed through the endotracheal tube to the larynx. The lidocaine desensitizes the larynx, and intubation usually proceeds uneventfully. The cuff should be inflated minimally, the pilot balloon should remain soft, and a leak around the cuff should occur at an inspiratory pressure of about 15 cm H_2O. The tube should be secured with rolled gauze behind the rabbit's ears. Alternatively, in large rabbits, a blind approach to endotracheal intubation is often successful when the head and neck are maximally extended and the endotracheal tube is advanced into the remiglottis. Intubation of the trachea is expedited at this point by listening for air movement through the tube while gently advancing it beyond the larynx into the trachea.

Fig. 18.30. Endotracheal intubation in a rabbit. The distal end of the endotracheal tube has been advanced through the larynx to its final position in the midcervical trachea. The anesthetist is preparing to extract the guide tube and secure the endotracheal tube behind the rabbit's ears.

Birds and Reptiles

Endotracheal intubation in birds and reptiles that are commonly presented for anesthesia is relatively easy. The glottis is usually located on the midline at the base of the tongue and is readily apparent when the patient's mouth is opened. Appropriately sized endotracheal tubes should be selected, and, to avoid damage to the tracheal rings if cuffed tubes are used, the cuff should not be overinflated. Owing to the small size of some birds and reptiles and the propensity for mucus to collect in the distal end of the tube, the anesthetist should be careful to assure a patent airway at all times. The use of lubricating jelly can also cause the obstruction of air flow through small endotracheal tubes.

Special Techniques for Endotracheal Intubation

The common techniques for endotracheal intubation may fail if oropharyngeal pathology is present (Fig. 18.31) or if movement of the temporomandibular joint is impaired. In such patients, "blind" intubation can be performed successfully on occasion. With the patient's head and neck extended, the larynx can be manipulated externally with one hand while the other hand maneuvers the tube through the larynx and into the trachea. However, if this technique is too traumatic or fails completely, other options for intubation are available.

Guide-Tube Technique

In some patients, a laryngoscope blade will allow exposure and illumination of the glottis by diverting the obstruction to one side, enabling direct placement of the endotracheal tube into the larynx. However, it may be easier to pass a small-diameter guide tube (e.g., a canine urinary catheter), rather than an endotracheal tube, through the glottis. Guide-tube technique has been previously described for various species, and it can be beneficial in dogs and cats with oropharyngeal pathology.[19] Once the tip of the guide tube is situated about half the distance from the cricoid cartilage to the thoracic inlet, the endotracheal tube can be passed into place (Fig. 18.32). Then, the guide tube is removed, and the endotracheal tube is secured as appropriate for the species involved.

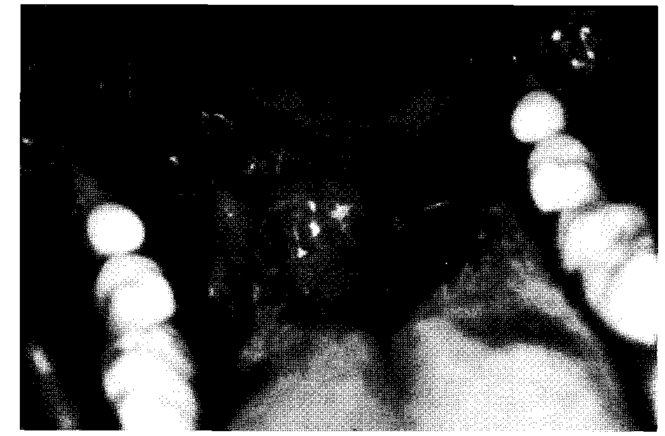


Fig. 18.31. In this dog, a pharyngeal tumor is obstructing the larynx and inhibiting passage of an endotracheal tube. Intubation was accomplished by using a laryngoscope blade to expose the glottis enough for a guide tube to enter the larynx, followed by passage of the endotracheal tube over the guide tube. From Hartsfield.[19]

Retrograde Intubation

If direct visualization of at least a portion of the glottis is impossible, other techniques of intubation have been advocated.[19,20] One method—use of a retrograde guide tube or wire—involves passing a hypodermic needle through the skin of the neck and into the trachea at the junction of the second and third tracheal rings. In human patients, the needle is passed through the cricothyroid membrane. A guidewire is then maneuvered through the needle cranially into the larynx, pharynx, and oral cavity until it can be used as a guide for passage of an endotracheal tube (Fig. 18.33). After the tip of the endotracheal tube is within the larynx, the needle and the guide tube are removed, and the endotracheal tube is manipulated into its final position with the cuffed end near the thoracic inlet. The cuff should be located caudal to the puncture site of the hypodermic needle to avoid forcing gases subcutaneously or into the mediastinum during positive-pressure ventilation. Subcutaneous emphysema and pneumothorax are possible complications with this technique.

Lateral Pharyngotomy

This technique has been described[6] and has been advocated for selected canine and feline patients requiring oropharyngeal surgery or orthopedic procedures involving the mandible or maxilla (Fig. 18.10). The major advantages are improved visualization within the operative field during oropharyngeal surgery and normal dental occlusion to aid in the proper reduction of mandibular or maxillary fractures.

The basics of tube placement involve passage of a correctly sized, cuffed endotracheal tube and a routine skin incision made near the angle of the mandible. Then, hemostats are bluntly passed through the skin incision into the caudal part of the pharynx. After the endotracheal tube adapter has been removed, the adapter end of the tube is grasped and pulled from the pharynx, through the subcutaneous tissue, and through the skin incision. The endotracheal tube adapter is replaced, and the tube is reconnected to the breathing system for maintenance. A correctly placed tube should be secured to the skin with tape and several sutures.

Using an Endoscope

Laryngoscopy with a flexible fiber-optic endoscope can be useful for intubation in patients with abnormal anatomy or disease

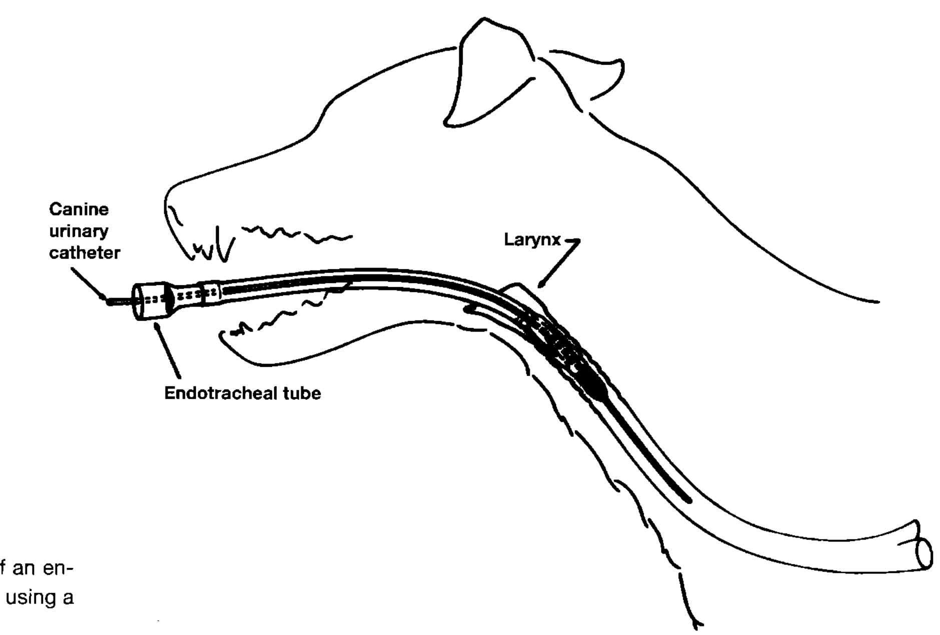


Fig. 18.32. Diagram illustrating passage of an endotracheal tube into the trachea of a dog by using a guide tube. From Hartsfield.[19]

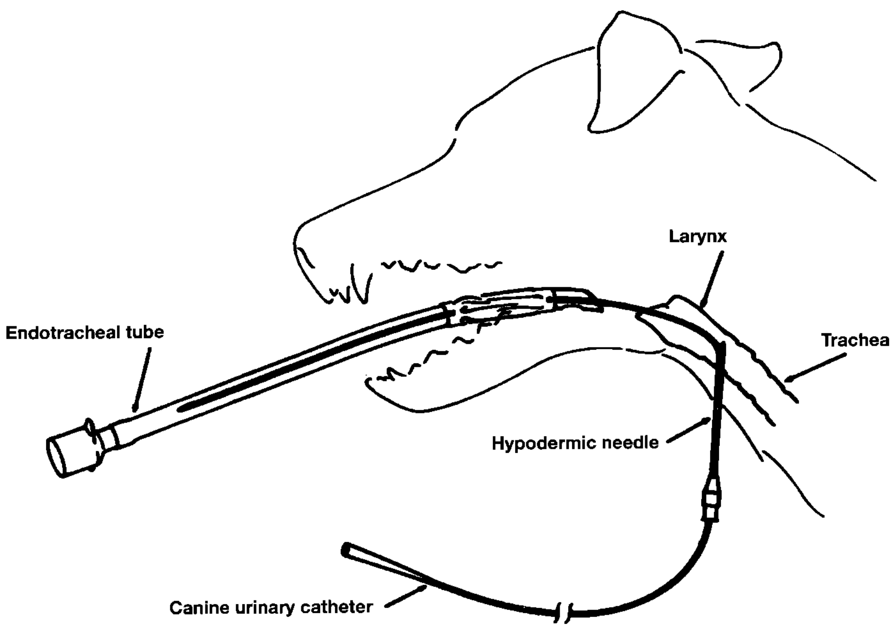


Fig. 18.33. Diagram illustrating the placement and use of a retrograde guide tube for passage of an endotracheal tube in a dog. This technique is reserved for patients that cannot be intubated by other methods. From Hartsfield.[19]

processes involving the pharynx or head and neck. Depending on the species and the specific conditions, the endoscope can be placed inside the endotracheal tube to directly guide intubation passed orally beside the endotracheal tube, or advanced through the nasal passage to view the endotracheal tube entering the glottis. The technique can be particularly advantageous in horses with abnormal oropharyngeal, laryngeal, and/or nasal anatomy, and can be helpful in small laboratory species that are difficult to intubate. The technique is applicable to any species that are difficult to intubate. The technique is applicable to any species in which intubation is impaired by anatomical abnormalities or disease. The technique is illustrated in a normal cat in Figs. 18.34 through 18.36.

Tracheostomy

A temporary tracheostomy can be chosen for airway management in lieu of the techniques suggested earlier for difficult cases. In some patients, the only reasonable option for intubation is tracheostomy, and some patients with airway disease arrive in the induction room with a tracheostomy tube in place. For anesthesia, intubation of the trachea through the tracheostomy site provides all of the advantages of oral intubation or intubation by pharyngotomy. However, tracheostomy has been associated with infection, granulomas, tracheal stricture, cartilage damage, hemorrhage, pneumothorax, tracheocutaneous or tracheo-esophageal fistula, aspiration, dysphagia, and tracheal malacia; thus, tracheostomy should not be considered an innocuous procedure.[21] Intubation via tracheostomy is generally reserved for patients requiring preoperative or postoperative tracheostomy for airway management. A tracheostomy tube with a replaceable lumen (Fig. 18.8) should be used, if available, but standard endotracheal tubes have been used satisfactorily (Fig. 18.9). Care of the tube is very important. Neglected tubes that are not cleaned regularly can be obstructed by mucus that dries within the lumen of the tube (Figs. 18.37 and 18.38).

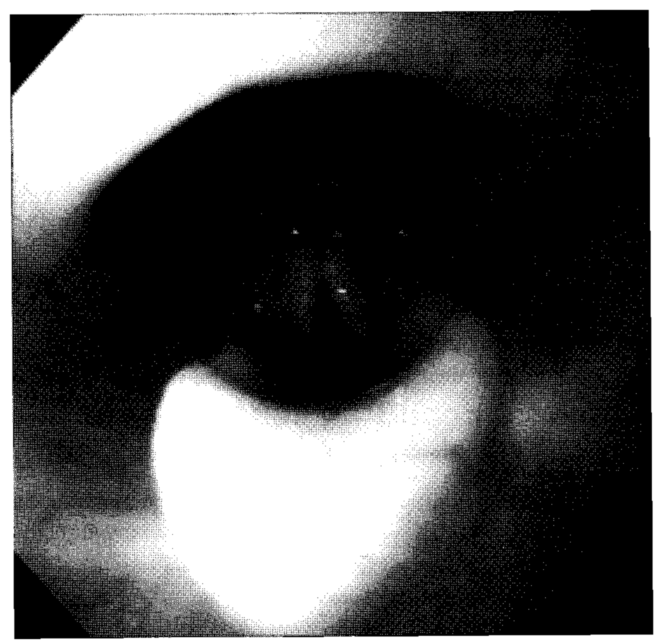


Fig. 18.34. Epiglottis, arytenoid cartilages, and glottis of a cat, as viewed through a fiber-optic endoscope.

Changing Endotracheal Tubes

Changing endotracheal tubes during a surgical or diagnostic procedure in an anesthetized animal is occasionally required due to a failing cuff or simply the need for a different size or length of tube. Patients positioned and draped for surgery are generally not ideally situated for intubation. Changing the tube with guide-tube technique is probably the easiest, most efficient way to accomplish the procedure.[19,22] Depending on the size of the patient and the endotracheal tube, two canine urinary catheters (8 to 10

Fig. 18.35. A polyethylene guide tube (8-French) passing through the glottis and into the larynx and trachea of a cat as viewed through a fiber-optic endoscope. Although not generally necessary for intubation in cats, this technique is effective in other species.

Fig. 18.36. An endotracheal tube passing into the larynx of a cat as viewed through a fiber-optic endoscope. Although not generally necessary for intubation in cats, this technique is effective in other species.

French) connected in tandem, or an equine stomach tube will make an excellent guide tube.

To change endotracheal tubes, the guide tube is inserted through the original endotracheal tube to the area of the midcervical trachea. Next, the endotracheal tube cuff is deflated, and the endotracheal tube is pulled over the guide tube without removing the guide tube from the trachea. Then, the new endotracheal tube is maneuvered through the larynx and into the trachea by using the guide tube to direct its passage. The cuff of the new tube is inflated to protect the airway, and the new tube is secured in the manner appropriate for the specific species.

Tracheal Extubation

Extubation is performed after patients regain the ability to swallow and protect their airways. When the cuff is deflated, the endotracheal tube is removed slowly and deliberately, with care taken to avoid damaging the patient's tissues with the endotracheal tube or damaging the cuff as the tube passes the teeth. After extubation, protection of the airway from foreign material and maintenance of a patent airway remain important. The type of surgical or diagnostic procedure, the species and breed, and preexisting conditions all affect these considerations.

The anesthetist should be certain that no foreign material remains in the oropharynx before beginning extubation. In dogs and cats, the pharynx should be inspected visually, and any debris should be removed. Specifically, surgery of the mouth and pharynx, dental procedures, and endoscopy promote the accumulation of blood, fluids, lubricants, tartar, or other materials. Animals anesthetized for gastrointestinal surgery are prone to passive movement of fluid into the pharynx; two examples are dogs with gastric dilation and volvulus (GDV) and horses with colic. Nasogastric or orogastric tubes commonly used in these procedures may promote flow of gastric contents into the pharynx during surgery or when the tube is removed. With either

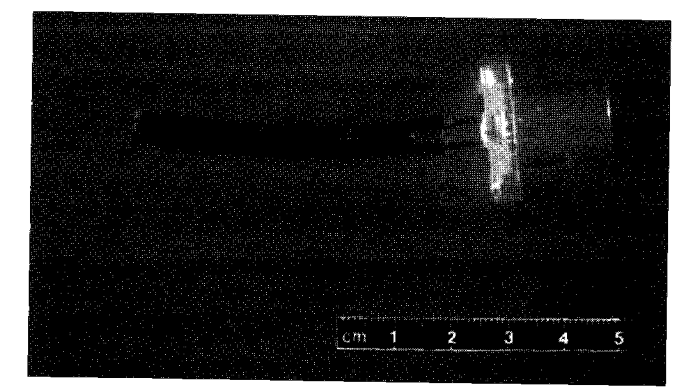


Fig. 18.37. An endotracheal tube that had been placed through a tracheostomy to maintain an airway in a cat during transport of the cat to a referral center. On presentation, the cat was dyspneic and cyanotic. The tube was filled with dried mucus, and the extent of the occlusion of the lumen is illustrated more dramatically in Fig. 18.38. The tube had not been changed or cleaned for several hours.

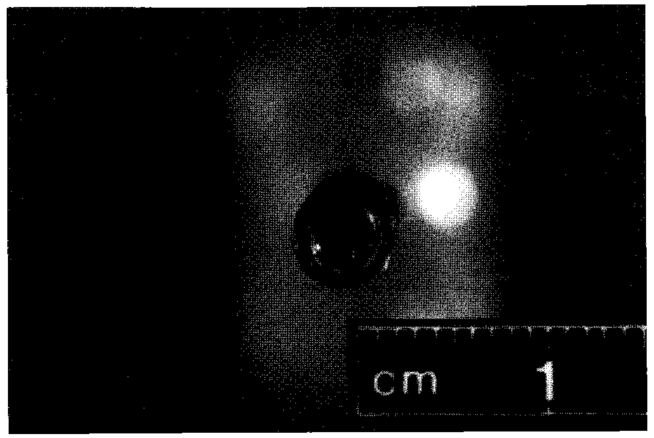


Fig. 18.38. End-on view of the endotracheal tube in Fig. 18.37 shows that the lumen was almost occluded. The cat was dyspneic prior to removal of the tube from the tracheostomy site.

species, the head should be positioned to allow drainage of fluid from the pharynx during surgery, and removing the endotracheal tube with the cuff inflated is advised.

Assuring a patent airway after extubation is essential, especially in patients with small-diameter upper airways (e.g., kittens, piglets, rabbits, and small brachycephalic dogs). A number of factors can be responsible for postextubation problems. Edema of the upper airway, including the larynx and nasal passages; laryngeal spasm; interference with the integrity of the airway by the soft palate; and laryngeal paralysis are all possible causes of obstructive problems. The anesthetist should be prepared to manage the airway at the time of extubation, knowing that these complications can impair ventilation and oxygenation. In some instances of postextubation airway obstruction, reanesthetizing the patient and reintubation may be the only feasible option.

Techniques of Oxygen Administration

Supplemental oxygen is used in anesthetized and critically ill patients to increase the partial pressure of oxygen in arterial blood (PaO_2) and to promote delivery of oxygen to the tissues. When a patient is breathing room air, values for PaO_2 that are less than 80 mm Hg indicate the potential for hypoxemia. If the PaO_2 decreases to less than 60 mm Hg, the need for supplemental oxygen is indicated.[23] Although ventilation is a factor in maintaining oxygenation, the fraction of oxygen in inspired gases (F_IO_2) plays a significant role in establishing the PaO_2. As a rule, the PaO_2 value is approximately five times the F_IO_2 value if there are no major abnormalities in the matching of pulmonary ventilation and perfusion. Supplemental oxygen may be the only effective way of correcting hypoxemia in animals with diffusion abnormalities and ventilation-perfusion mismatching. Supplemental oxygen may not significantly improve PaO_2 in patients with pulmonary or cardiac shunts.

Several techniques can be used to administer oxygen to anesthetized and critically ill patients. The effectiveness of oxygen supplementation is assessed by evaluation of the patient's clinical responses (e.g., improvement in mucous membrane color and character of ventilation), by measuring the F_IO_2, and by monitoring of PaO_2, arterial oxygen saturation (SaO_2), and saturation of peripheral oxygen (SpO_2). Although PaO_2 and SaO_2 data are reliable, they require periodic arterial blood sampling and the use of an acid-base, blood-gas analyzer. The SpO_2 can be conveniently measured by pulse oximetry. Pulse oximetry is a practical method for noninvasive, moment-to-moment estimation of the saturation of hemoglobin with oxygen in anesthetized, recovering, and critically ill patients.[24,25]

Mask Delivery

Masks for delivery of oxygen to veterinary patients are useful for preoxygenation immediately before induction of anesthesia and for emergency situations in awake patients. The use of masks for oxygenation requires constant attention, and some patients will not accept a mask unless they are sedated. Both factors limit the effectiveness of masks in awake patients. Indeed, some patients object to a mask so vigorously that the increase in oxygen consumption associated with restraint may nullify the benefits of a greater F_IO_2.

The flow rates generally recommended for increasing F_IO_2 when using masks are variable among species. For example, flow rates of 10 to 15 L/min of supplemental oxygen have been recommended to increase the inspired-oxygen concentration to approximately 35% to 60% in adult horses.[26] Flow rates for smaller patients, including dogs and cats, usually range from 3 to 5 L/min. With a tight-fitting mask, higher flow rates of oxygen will produce greater F_IO_2 values and less rebreathing of expired carbon dioxide.

A mask should be used with a breathing system with a reservoir that can meet the patient's tidal volume demands or with a valved system that allows room air to be entrained. As an example, a dog with a tidal volume of 300 mL and an inspiratory time of 1 s has a peak inspiratory gas flow of approximately 18 L/min, which exceeds the practical flow rate for oxygen during masking. High inspiratory flow rates can be provided if the mask is attached to a circle breathing system with a reservoir bag. In addition, a breathing system has an overflow (pop-off) valve that prevents the buildup of excessive pressure with a tight-fitting mask.

Nasal Insufflation

Insufflation involves delivery of oxygen into the patient's airway at relatively high flow rates (Fig. 18.39); the patient inspires both oxygen and room air, the relative proportions of each being determined primarily by the oxygen flow rate and the rate of gas flow during inspiration.

Insufflation can be accomplished by a variety of methods. For horses recovering from anesthesia, oxygen may be delivered from a flowmeter through a delivery tube and into an orotracheal, nasotracheal, or tracheostomy tube. For most awake patients, oxygen is insufflated through a nasal catheter, the tip of which is positioned in the nasopharynx. The catheter is usually made of soft rubber, and the tube should have several fenestrations to pre-

Fig. 18.39. A nasal catheter for administration of oxygen in a dogs. The tube is secured to the muzzle with a suture.

vent jetting lesions from developing in the nasopharyngeal mucosa.[27] For awake small animals, instilling 2% lidocaine into the nasal passage with the patient's head and neck extended and held upward may facilitate passage of the tube. Placement involves insertion of the rubber catheter into the nasal passage and the nasopharynx, the distance being approximately the same as from the tip of the nose to the medial canthus of the eye. The external portion of the catheter is secured to the patient's head with tissue adhesive, tape, and/or sutures. A flexible length of tubing supplies oxygen from a flowmeter and allows the patient some freedom for movement in a cage or stall. Changing the catheter to the opposite nasal passage every 1 to 2 days has been recommended to prevent pressure necrosis, jet lesions, and accumulation of mucus.[27]

The flow-rate requirements for oxygen during insufflation are quite variable, the patient's ventilation and the desired F_IO_2 being two important factors. Following anesthesia, adult horses require a minimum of 15 L/min of oxygen flow to improve the PaO_2 in arterial blood, and proportionally lower flows (e.g., 5 L/min) are suitable for smaller horses and foals.[26] In small animals, flow rates of 1 to 7 L/min are typically used for the administration of nasal oxygen. Approximate flow rates for dogs and cats to achieve rather specific ranges of F_IO_2 have been suggested.[27,28] In dogs, various flow rates of 100% oxygen administered intranasally were studied, and flow rates of 50, 100, 150, and 200 mL kg^{-1} min^{-1} produced inspired-oxygen concentrations measured at the tracheal bifurcation of 28%, 37%, 40%, and 47%, respectively.[29] To prevent mucosal drying with prolonged insufflation, oxygen should be flowed through a bubble-type humidifier.

Tracheal Insufflation

An intratracheal catheter placed percutaneously into the trachea through the cricothyroid membrane or between tracheal rings near the larynx can be used to insufflate oxygen to a compromised patient. Intratracheal administration of 100% oxygen has been evaluated in dogs, and flow rates of 10, 25, 50, 100, 150, 200, and 250 mL kg^{-1} min^{-1} produced inspired-oxygen concentrations at the tracheal bifurcation of 25%, 32%, 47%, 67%, 70%, 78%, and 86%, respectively.[30] The technique for tracheal insufflation has been described for small animals.[27,30] The catheter should be placed aseptically, be of the over-the-needle type, relatively large bore, have several smooth fenestrations to prevent jet lesions, and ultimately positioned with the tip near the bronchial bifurcation. Oxygen should be humidified, and flow rates should approximate those used for nasal insufflation.

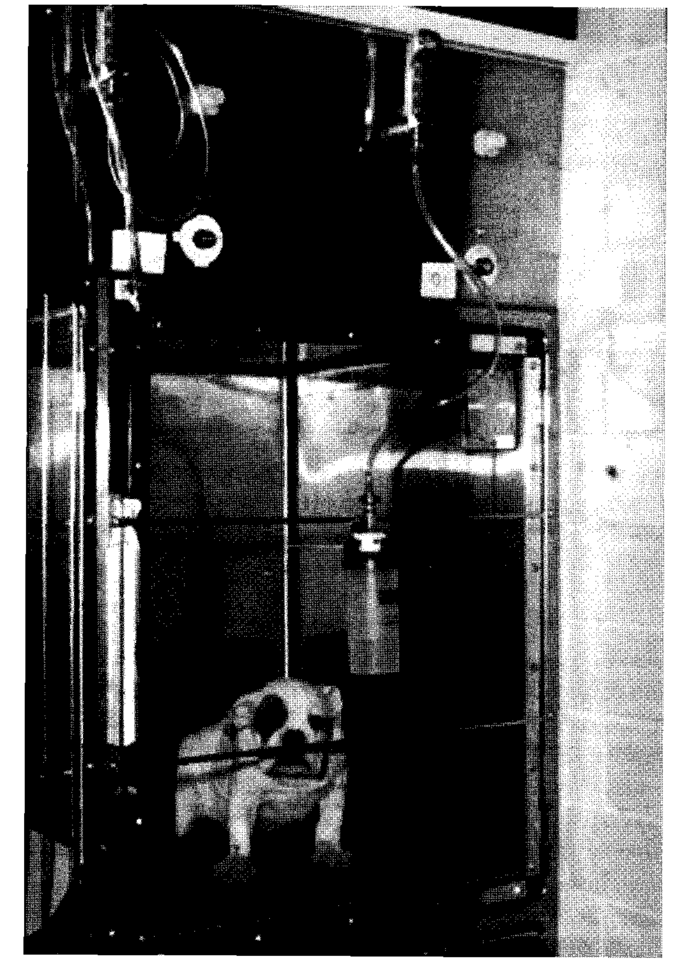


Fig. 18.40. A standard stainless-steel cage with a Plexiglas door facilitating administration of humidified oxygen to an English bulldog. Although concentrations of oxygen are unlikely to be very high, patients with respiratory distress often show clinical improvement.

Oxygen Cages

Oxygen cages (Figs. 18.40 and 18.41) specifically designed for small animals are commercially available, but expensive. These cages regulate oxygen flow, control humidity and temperature, and eliminate carbon dioxide from exhaled gases. For small animals, flow rates of oxygen, cage temperature, and cage humidity have been recommended to be less than 10 L/min, approximately 22°C, and 40% to 50%, respectively.[27] With these flow rates,

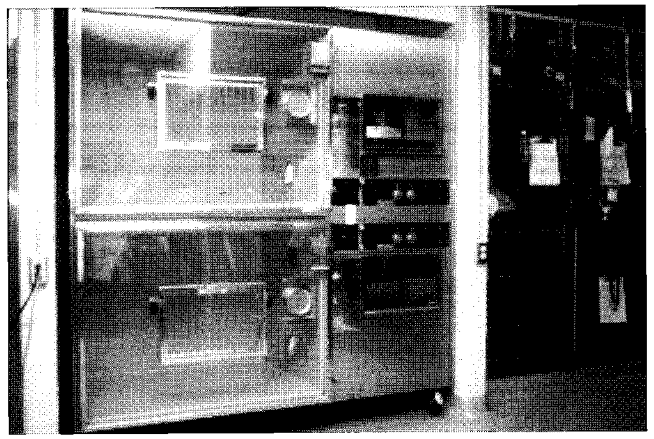


Fig. 18.41. Two commercial oxygen cages designed for small veterinary patients. The cages can control environmental temperature, deliver oxygen, and absorb carbon dioxide from expired gases.

most oxygen cages produce an environmental oxygen concentration of about 40% to 50%.[27] Oxygen concentrations of 30% to 40% generally are adequate for patients with moderate pulmonary disease.[23] Oxygen cages are not practical for large horses,[26] and, even in smaller animals, the effectiveness of an oxygen cage diminishes as body size increases. Because of this, nasal insufflation of oxygen has supplanted oxygen cages in many instances, even for smaller dogs and cats.

Smaller canine patients can be managed easily in oxygen cages, but temperature and humidity are more difficult to control with larger dogs. A major disadvantage of an oxygen cage is that the animal must be removed from the cage for examination and treatment, requiring the patient to breathe room air or oxygen by mask during this period. Clinically, some dogs and cats with serious ventilatory compromise respond very well to an oxygen-enriched environment as initial therapy; the increase in F_IO_2 is associated with decreased ventilatory effort, and the patient stabilizes and becomes more manageable prior to further examination and treatment.

Oxygen Toxicity

Oxygen toxicity develops with prolonged exposure to high oxygen concentrations.[31,32] Oxygen toxicity leads to the deterioration of pulmonary function, pulmonary edema, and death. The length of time that a patient's PaO_2 is elevated may be more predictive of oxygen toxicity than the duration of exposure to a high F_IO_2.[32] There is significant species and individual variability in susceptibility to oxygen toxicity.[27,32] In human patients, the guideline is that 100% oxygen should not be administered for more than 12 h of exposure.[27] In general, a patient should not be deprived of a high concentration of oxygen if a high F_IO_2 is required to maintain an adequate PaO_2; it has been stated that the brain softens (due to hypoxemia) before the lungs harden (owing to changes induced by prolonged exposure to high oxygen tensions).[33] As a guideline for prolonged administration of oxygen, 40% to 50% oxygen is generally safe, but higher inspired concentrations should be used if necessary to maintain a patient's PaO_2 at approximately 90 to 100 mm Hg and ensure hemoglobin saturation with oxygen.

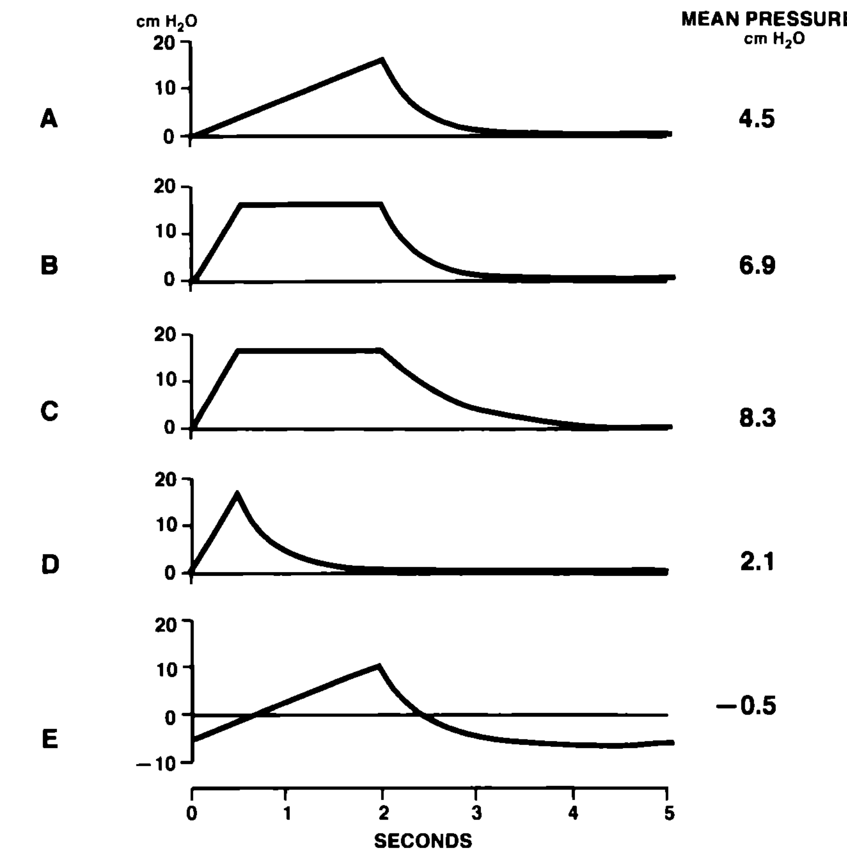
Mechanical Ventilation

Essentially, all anesthetized patients hypoventilate; they do not maintain arterial carbon dioxide partial pressure ($PaCO_2$) values near 40 mm Hg because of abnormal alveolar ventilation. Although controlled ventilation is not necessary for all anesthetized patients, various circumstances may compel an anesthetist to employ intermittent positive-pressure ventilation (IPPV). The absolute indication for mechanical ventilation is apnea.[34] However, IPPV should be instituted if hypoventilation becomes significant, if neuromuscular blocking drugs are employed, or if intrathoracic surgery is performed.[35] General anesthesia for longer than 11/2 h may constitute a reason for IPPV.[36] In addition, IPPV may be needed to facilitate inhalant anesthesia. Hypoventilating animals may not absorb enough anesthetic to maintain surgical anesthesia. IPPV will enhance alveolar ventilation and increase the uptake of the inhalants. This may help in eliminating the oscillations between deep and light anesthesia associated with depression of spontaneous ventilation.

Maintaining relatively normal carbon dioxide tensions in arterial blood is the primary goal of mechanical ventilation in anesthetized patients. Normal values for $PaCO_2$ are generally in the range of 35 to 45 mm Hg for most species. However, controversy exists about the routine use of IPPV in anesthetized patients, particularly anesthetized horses, simply to keep $PaCO_2$ near 40 mm Hg. Because IPPV is associated with reduced cardiovascular function and moderate increases in $PaCO_2$ are associated with improvement in some cardiovascular variables, IPPV for every anesthetized patient is neither universally accepted nor universally practiced. The definition of an acceptable degree of hypercapnia in an anesthetized animal is not without debate. For spontaneously breathing horses, a range of 60 to 70 mm Hg of $PaCO_2$ has been suggested as perhaps safer than controlled ventilation with its potentially adverse effects on cardiovascular function and tissue perfusion.[36] Others have recommended IPPV for horses only when $PaCO_2$ values exceed 60 mm Hg[37] or even 70 mm Hg.[26,37] Employing IPPV in large anesthetized animals, including horses and other species, to maintain $PaCO_2$ values between 35 and 50 mm Hg remains common practice. For a more definitive answer to this debate, large-scale clinical trials with various sized horses and anesthetic-surgical conditions need to be completed.

The direct cardiovascular effects of carbon dioxide include dilation of peripheral arterioles and myocardial depression. Indirectly, carbon dioxide evokes sympathoadrenal responses, which cause blood pressure elevation, tachycardia, and increased myocardial contractility.[38] Moderate (60 to 70 mm Hg) to high (75 to 85 mm Hg) increases in $PaCO_2$ in spontaneously breathing and mechanically ventilated, lightly anesthetized horses were associated with augmented cardiovascular function compared with horses with normal carbon dioxide tensions; these hemodynamic effects were accompanied by increases in circulating catecholamines.[39] If more normal (35 to 45 mm Hg) arterial carbon

Fig. 18.42. Diagrams illustrating the mean pressure in the lungs in relation to positive-pressure ventilation: the effects of a long inspiratory time **(A)**, an inspiratory plateau or holding pressure at the end of inspiration **(B)**, an inspiratory plateau with retarded expiration **(C)**, a rapid inspiration **(D)**, and a slow inspiration with a negative expiratory phase **(E)**. Lower mean pressure is most desirable from the standpoint of cardiovascular function, and **D** illustrates the most desirable type of ventilation, although the inspiratory time depicted is shorter than normally used clinically. From Mushin et al.,[71] in Lumb and Jones.[72]



dioxide tensions correlate with lower values for blood pressure, myocardial contractility, and cardiac output in anesthetized animals, the argument can be made that spontaneously breathing (slightly hypercapnic) anesthetized animals may maintain cardiovascular function better than animals whose ventilation is controlled with IPPV. The use of inotropic drugs may be necessary to maintain cardiovascular function during surgical anesthesia in mechanically ventilated horses. Nevertheless, there are good reasons to maintain $PaCO_2$ values within reasonable limits in anesthetized animals.

Inhalant anesthetics (e.g., halothane) and epidural or spinal anesthesia reduce the circulatory responses to carbon dioxide.[38,40] In addition, hypercapnia has been associated with increases in vagal tone and slowing of heart rate.[41] Indeed, hypercapnia has long been related to enhanced vagal responsiveness, bradycardia, and even cardiac arrest. It used to be said that "hypercarbia does not stimulate vagal activity directly, but it 'sets the stage' for cardiac arrest if such a stimulus is present."[42] It is known that carbon dioxide produces narcosis in dogs, the degree of which depends on the $PaCO_2$ value; narcosis progressively increases with $PaCO_2$ values above 95 mm Hg and induces complete anesthesia at 245 mm Hg.[43] Hypercapnia and the associated increases in circulating catecholamines have been linked to the development of cardiac arrhythmias, especially when the heart has been sensitized by halogenated inhalant anesthetics.[44] In human pediatric patients whose airways were managed by masks, hypercapnia was associated with an increased incidence of arrhythmias.[45] The authors noted that light anesthesia or a combination of factors such as hypercapnia and halothane might have been as important as hypercapnia alone in the production of arrhythmias in these children. Thus, there is a negative side to uncontrolled hypercapnia that should be considered for anesthetized patients, and the veterinarian or anesthesiologist should weigh the advantages against the disadvantages in the anesthetic management of each patient.

Mechanical ventilation does negatively affect cardiovascular function (Fig. 18.42). The depression of cardiovascular function may be significant. When ventilation is controlled and negative pressures are not generated during inspiration, venous return is not enhanced. Indeed, IPPV may physically impede venous return to the right side of the heart, leading to decreases in stroke volume, cardiac output, and arterial blood pressure. In anesthetized, mechanically ventilated horses, a reduction in blood pressure and damping of the pressure waveform is not uncommon, especially in critically ill patients with a marginal blood volume. The negative effects of mechanical ventilation on cardiovascular function can be exacerbated by prolonging inspiratory time, holding positive pressure in the lungs at the end of inspiration, retarding exhalation, applying positive pressure during the expiratory phase, and employing an excessively rapid respiratory rate. Some of these effects are illustrated in Fig. 18.42. Fortunately, these negative effects can be overcome in many cases by the appropriate expansion of extracellular fluid volume and, if necessary, the administration of inotropic drugs.

Guidelines for mechanical ventilation usually include values for inspiratory time, respiratory rate, inspiratory to expiratory time ratio, and tidal volume. Some variations exist because of differences in body size, species, physical condition of the lungs and thorax, and existing disease processes.

Normal tidal volume is generally considered to range between 10 and 20 mL/kg of body weight.[46] A good working guideline for tidal volume in the domestic species is approximately 10 mL/kg.[47] For IPPV, the tidal volume set on a mechanical ventilator is usually increased above the normal spontaneous tidal volume to compensate for pressure-mediated increases in the volume of the breathing system and airway. Increasing the tidal volume (bellows volume) by 2.2 to 4.4 mL/kg has been recommended.[35] Settings for tidal volume—15 mL/kg in large animals and 20 mL/kg in small animals—have been suggested.[47] Use of small tidal volumes may cause atelectasis, which may only be recognized grossly during thoracotomy or with blood-gas analysis, because atelectasis contributes to mismatching of pulmonary ventilation and perfusion leading to decreased PaO_2. The tidal volume should be delivered to the patient over a relatively short period to avoid maintaining positive intrathoracic pressure. Inspiratory time should be approximately 1 to 1.5 s in small animals and 1.5 to 3 s in large animals.

The inspiratory time compared with time during the entire expiratory phase is termed the *I-E ratio*. That fraction should be 1:2 (I-E) or less for mechanical ventilation in all patients. Ratios that approach 1:1 produce a long duration of positive intrathoracic pressure, which interferes more with cardiovascular function. If a patient's respiratory rate is 10 breaths/min and the inspiratory time is 1.5 s, the I-E ratio will be 1:3. In general, the exact value of the I-E ratio is not important as long as the ratio is less than 1:2. With some ventilators that incorporate specific, unchangeable I-E ratios, the options for controlling respiratory rate may be limited.

Tidal volume and inspiratory time affect the development of peak inspiratory pressure. In general, 15 to 30 cm H_2O will expand the lung,[47] and 15 to 20 cm H_2O and 20 to 30 cm H_2O have been recommended as peak inspiratory pressures for mechanical ventilation of small animal species with normal lungs and large animal species with normal lungs, respectively.[35] Excessive or sustained pressure during IPPV can cause excessive expansion and volutrauma, leading to disruption of the alveolar membrane, to the development of interstitial air, and ultimately to the transfer of air into the mediastinum, pleural space, or abdomen.[48] A good guideline for peak inspiratory pressure is not to exceed 30 cm H_2O. Special attention to peak pressure is important for animals that have experienced lung trauma (e.g., diaphragmatic hernia).[49]

The appropriate respiratory rate for mechanical ventilation varies with the species and the tidal volume selected. The following recommendations have been published: dogs, 8 to 14 breaths/min; cats, 10 to 14 breaths/min; horses and cows, 6 to 10 breaths/min; and small ruminants and pigs, 8 to 12 breaths/min.[47] In patients requiring smaller than usual tidal volumes, to avoid excessive inspiratory pressures (e.g., lung trauma, diaphragmatic hernia, or gastrointestinal distension, including GDV), respiratory rates can be increased to maintain the appropriate minute ventilation.

When controlled ventilation is discontinued at the end of anesthesia, the return of spontaneous ventilation may be impaired. If $PaCO_2$ is low, spontaneous ventilation may not resume. Part of the management of controlled ventilation should be maintenance of relatively normal carbon dioxide tensions, and hypocarbia should be avoided. The residual effects of opioids, anesthetics, and adjunctive drugs (e.g., neuromuscular blocking drugs) at the end of anesthesia may contribute to a delayed return to spontaneous ventilation. Complicating factors associated with general anesthesia or surgery (e.g., hypothermia or hypovolemia) may slow an animal's return to consciousness and thus spontaneous ventilation. In general, the arterial tension of carbon dioxide must increase to stimulate the animal to breathe spontaneously, or the patient must regain a level of consciousness that promotes spontaneous ventilation. Before attempting to discontinue controlled ventilation, the anesthetist should ascertain that depth of anesthesia is decreasing, that the effects of muscle-relaxing drugs have subsided or have been antagonized, and that cardiovascular function is relatively normal. Then, the animal can be weaned from controlled ventilation.

The patient should continue to receive supplemental oxygen until spontaneous ventilation is relatively normal. Reducing the rate of controlled ventilation usually increases $PaCO_2$ enough to stimulate spontaneous breathing when an animal is regaining consciousness. Generally, the patient is mechanically or manually ventilated at a rate of one to four breaths per minute until spontaneous ventilation resumes. In many cases, animals begin to breathe spontaneously after the vaporizer has been turned off and most of the inhalant anesthetic has been eliminated, even though controlled ventilation has not been stopped. Some patients require some type of external stimulus to begin breathing (e.g., pinching the skin between a dog's toes). After the animal begins to breathe spontaneously, assisted ventilation and supplemental oxygen should be provided until the respiratory rate and tidal volume begin to normalize.

Anesthesia Ventilators

Anesthesia ventilators provide for mechanical ventilation of patients being maintained with inhalant anesthetics. Simply, an anesthesia ventilator is a reservoir bag (a bellows or concertina bag) in a closed container (bellows housing) that can substitute for the reservoir bag of an anesthesia breathing system. Within limits, the ventilator can drive its bellows to produce a specific tidal volume or a specific inspiratory pressure at a preselected rate. The anesthesia ventilator performs the same job as the anesthetist who periodically squeezes the circle system's reservoir bag to ventilate the patient. Some anesthesia ventilators are stand-alone units that are attached to an anesthesia machine when needed, whereas other ventilators are manufactured as an integral part of the anesthesia machine. Most anesthesia ventilators designed for human patients are appropriate for veterinary patients weighing less than approximately 140 kg. Ventilators specifically designed for large animals are needed for patients weighing more than 140 kg. Admittedly, these guidelines for body weight and selection of a ventilator are somewhat arbitrary.

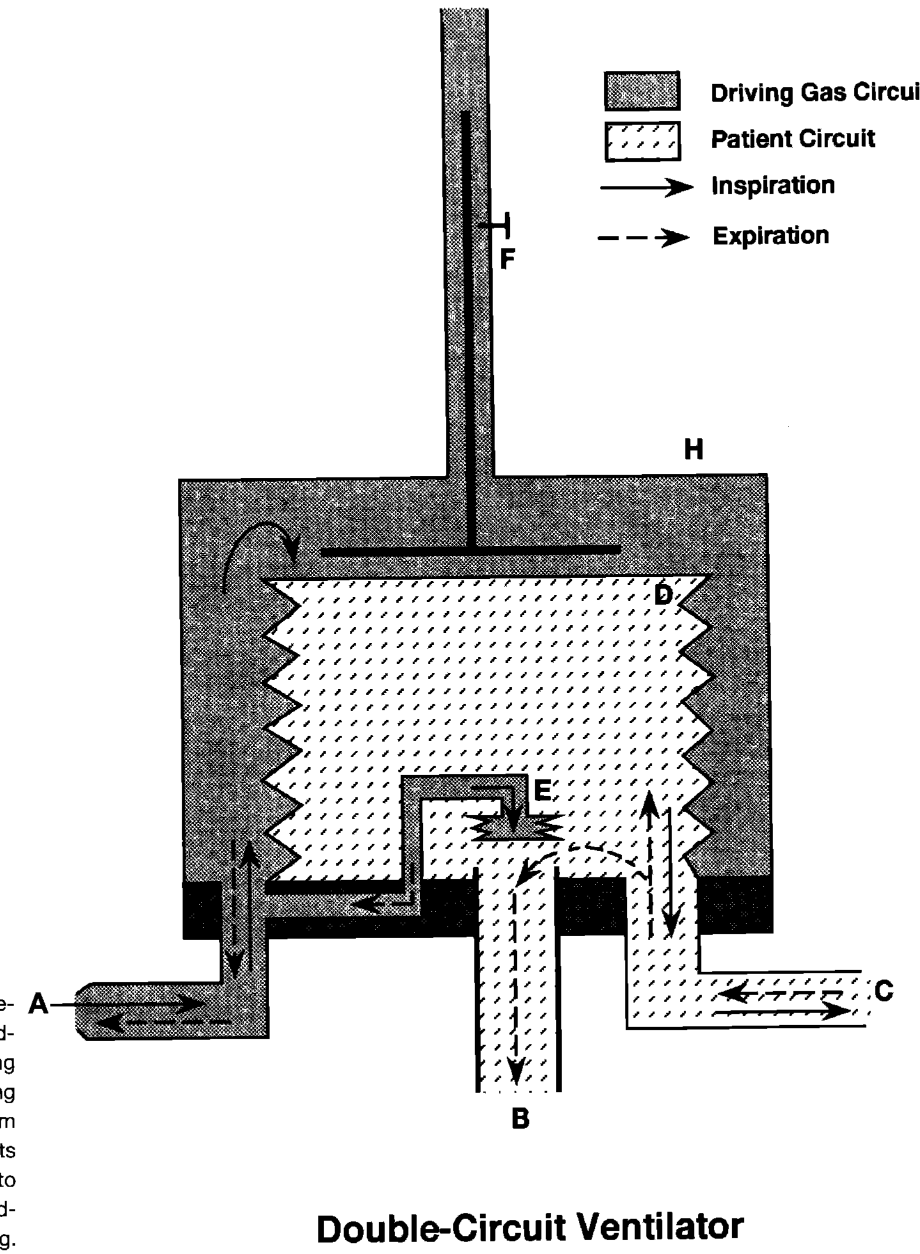


Fig. 18.43. Diagram of a generic, double-circuit ventilator. Driving gas enters at *A*, leading to compression of the bellows and forcing gas in the patient circuit toward the breathing system and the patient's respiratory system (*C*). Overflow gas from the patient circuit exits through the pop-off valve (*E*) and flows into the scavenger system (*B*). *F*, tidal volume adjustment; *D*, bellows; and *H*, bellows housing.

Classification

The power source, drive mechanism, cycling mechanism, and type of bellows have been used to classify anesthesia ventilators.[50] The power source may be electricity, compressed gas, or both. The drive mechanism is commonly compressed gas, even when electric controls are used. Anesthesia ventilators are usually double-circuit units (Fig. 18.43). *Double circuit* refers to two gas sources: (a) the driving-gas circuit (outside of the bellows), which compresses the bellows, and (b) the patient gas circuit (inside the bellows), which originates at the anesthesia machine and provides oxygen and anesthetic to the breathing system and patient. Specific ventilators for veterinary applications are classified in Table 18.1.

Anesthesia ventilators are typically, though not always, time cycled.[50] Fluidic timing devices were common in the late 1970s, and fluid-controlled ventilators remain in use. Newer electronic ventilators incorporate solid-state timing circuitry and are classified as time cycled and electronically controlled. A pressure-cycling mechanism may be present in some ventilators, and some have been described as volume cycled. In most cases, a timing mechanism plays a major role in a ventilator's function, and volume or pressure limits may affect the change in the respiratory cycle from inspiration to expiration.

The direction that the bellows moves during expiration, either ascending or descending, also helps to characterize anesthesia ventilators. Newer anesthesia ventilators usually have ascending

Table 18.1. Classification and characteristics of some anesthesia ventilators

Ventilator	Power Source	Drive Mechanism	Cycling Mechanism	Bellows[a]	Type of Ventilation
Drager SAV (SA)	Pneumatic	Pneumatic	Time-fluidic	Ascend	Control
Hallowell EMC 2000 (SA)	Pneumatic and electronic	Pneumatic	Time-electronic	Ascend	Control
Mallard 2400 V (SA)	Pneumatic and electronic	Pneumatic	Time-electronic	Ascend	Control
Metomatic (Ohio)-SA	Pneumatic	Pneumatic	Time-fluidic	Descend	Assist/control
Ohmeda 7000 (SA)	Pneumatic and electronic	Pneumatic	Time-electronic	Ascend	Control
ADS 1000 (SA)	Pneumatic and electronic	Pneumatic	Time-electronic	None	Control
SAV 75 (SA)	Pneumatic	Pneumatic	Time-pressure	Ascend	Assist/control
Drager AV (LA)	Pneumatic and electronic	Pneumatic	Time-fluidic	Descend	Control
Narkovet Electronic LA Control Center	Pneumatic and electronic	Pneumatic	Time-electronic	Descend	Control
LAVC 2000 (LA)	Pneumatic	Pneumatic	Time-pressure	Descend	Assist/control
Mallard 2800 (LA)	Pneumatic and electronic	Pneumatic	Time-electronic	Ascend	Control

LA (large animal) and SA (small animal) indicate the primary use of the ventilator.
[a]Bellows is described in reference to the direction of movement during expiration.

bellows. The ascending bellows is considered safer because it will not fill if a disconnection occurs in the breathing circuit.[50] The ascending bellows falls to the bottom of the bellows housing during a disconnection, giving an immediate visual indication of ventilator failure. Ascending bellows are incorporated into modern electronic ventilators. Ventilators with descending bellows may continue to cycle even with a complete disconnection of the ventilator from the breathing system.

The terms *tidal volume preset*, *volume preset*, and *volume constant* have been used to describe anesthesia ventilators; these terms have been included in operation manuals and other descriptive literature authored both by manufacturers and by medical personnel. The implication is that the ventilator delivers exactly the tidal volume selected despite the total inspiratory time or the amount of inspiratory pressure that develops. However, the tidal volume that actually reaches a patient's lungs may vary from the setting on the ventilator. Variations are related to the compliance of the breathing system, leaks in the system, and the entry of fresh gases into the breathing system. Although the terms *tidal volume preset*, *volume preset*, and *volume constant* may be practical, the user should understand the unknown influences on the quantity of gas actually delivered to the patient.

In addition, ventilators may be called *pressure preset*, indicating that inspiration continues until a selected pressure is reached, no matter what tidal volume is necessary to achieve the pressure. The amount of gas delivered to a patient depends on a number of factors, including the resistance and compliance of the breathing system and the patient's respiratory system. Although inspiratory pressure may not vary over time, the tidal volume may change as compliance of the respiratory system changes.

Even though ventilators have been classified as *volume limited* and *pressure limited*, and *pressure cycled*, *time cycled*, and *volume cycled*, these terms may be somewhat misleading or confusing because the mechanism that actually causes the change from expiration to inspiration is most often a timing mechanism. Indeed, the change from one phase of ventilation to the other may involve more than one mechanism, including volume, pressure, and/or time. Some ventilators may use different cycling mechanisms depending on the mode of operation.[50] A pressure-limited ventilator is one that delivers gas to a patient during inspiration until a preset pressure develops in the bellows, at which point the expiratory phase begins. The disadvantage of pressure-limited ventilation is that the tidal volume delivered to a patient may decrease if respiratory compliance decreases during ventilation. A volume-limited ventilator delivers a present tidal volume (within the limits discussed earlier) without regard for the maximum inspiratory pressure (up to the preset maximum pressure for the ventilator). Inspiratory pressure may increase if compliance decreases during mechanical ventilation. Most anesthesia ventilators have a maximum pressure limit during inspiration for the safety of the patient, and that pressure limit varies with the model of the ventilator. Ventilators that are used as volume-limited units may truly be limited by time rather than by volume, and that fact becomes apparent if the inspiratory flow rate is too slow.

Terminology of Mechanical Ventilation

Several abbreviations are used in the medical literature to describe various types of ventilation. Common abbreviations are included and discussed in the following subsections.

IPPV (Intermittent Positive-Pressure Ventilation)

With IPPV, airway pressure is maintained above ambient pressure during inspiration, and airway pressure falls to ambient pressure to allow passive expiration.[48] *Conventional positive-pressure ventilation* (CPPV), also called *control-mode ventilation* (CMV), is a form of IPPV in which a ventilator delivers a preset tidal volume at a preset frequency.[51] *Assist-control-mode ventilation* (AMV) provides a preset tidal volume from the ventilator in response to patient-initiated attempts to inspire; a preset frequency of ventilation is delivered by the ventilator if the patient fails to initiate breathing.[51] The term *intermittent positive-pressure breathing* (IPPB) is synonymous with IPPV.

PEEP (Positive End-Expiratory Pressure)

With PEEP, airway pressure at end expiration is maintained above ambient pressure. The term *PEEP* is applied when positive pressure is maintained between inspirations that are delivered by a ventilator.[51] The term *ZEEP*, or *zero end-expiratory pressure*, has been used in studies comparing the effects of PEEP with the effects of ZEEP.[52] In addition, some ventilators can create negative pressure to assist expiration or to speed the egress of gases during the expiratory phase. This has been termed *NEEP* or *negative end-expiratory pressure*.

CPAP (Continuous Positive Airway Pressure)

When airway pressure is maintained above ambient pressure during spontaneous breathing, the term *CPAP* is applied instead of PEEP.[51]

IMV (Intermittent Mandatory Ventilation)

This method of ventilation is used for ventilatory support and for weaning of patients from ventilators. The technique allows patients to breathe spontaneously, but it inserts mechanical breaths at a preset tidal volume and frequency.[51,52] Most veterinary anesthesia ventilators are not designed for delivering this form of ventilation, and IMV is provided by critical care ventilators for human patients. The periodic sigh that anesthetists provide manually during spontaneous ventilation to expand the lung and decrease collapsed alveoli in anesthetized animals may be considered IMV.[26]

The terms *assisted ventilation* and *controlled ventilation* are common in the veterinary literature. Assisted ventilation can be performed manually by anesthetists, who synchronizes their compression of the breathing bag with a patient's spontaneous breathing to augment the tidal volume.[53] With a mechanical ventilator, assisted ventilation is basically patient-initiated ventilation, with the ventilator delivering the preselected tidal volume. Since the patient determines the frequency of ventilation, it also determines minute volume.[54] Some veterinary ventilators can provide assisted ventilation, delivering a tidal volume from the bellows when a patient creates negative pressure at the initiation of a breath.[55]

During *controlled ventilation* as defined earlier for CMV, inspiration is initiated by the ventilator and a preset respiratory rate is maintained. The ventilator sets frequency, tidal volume, and minute volume. Controlled ventilation is necessary in any situation that renders a patient unable to initiate an adequate number of breaths. Essentially all anesthesia ventilators will operate in this mode, and some operate only in this mode. Controlled ventilation can be provided manually by an anesthetist using the reservoir bag of the breathing system to establish both rate and tidal volume, and thus minute volume, for the patient.[53]

The term *assisted-controlled ventilation* has been defined as assisted ventilation (patient-controlled rate with ventilator-controlled tidal volume) with a preset minimum acceptable respiratory rate; if the patient-initiated rate falls below the preset rate, the ventilator will cycle at the minimum preset rate. This mode is similar to AMV as defined earlier. The use of assisted-controlled ventilation has been suggested for the transition period between spontaneous and controlled ventilation.

Guidelines for Use

The controls on most anesthesia ventilators include settings for tidal volume, inspiratory time, inspiratory pressure, respiratory rate, and I-E ratio (either adjustable or preset). Other controls may be present, but the four listed are basic. The following guidelines have already been discussed, but will again be briefly reviewed. The setting for tidal volume is usually between 10 and 20 mL/kg, and the inspiratory pressure is normally between 12 and 30 cm H_2O. In small patients, the respiratory rate should be set between 8 and 12 breaths/min, whereas the respiratory rate for large animals should be set between 6 and 10 breaths/min. In setting the ventilator, inspiratory time should be short in comparison to expiratory time so that positive interpleural pressure will minimally interfere with venous return and cardiac output. Inspiratory time should be 1 to 1.5 s in small animals and preferably less than 3 s in large animals. Therefore, the I-E ratio should be 1:2 or less (e.g., 1:3 or 1:4), depending on the respiratory rate.

Examples of Ventilators for Small Animals

Although not all-inclusive, the following discussion describes ventilators that are appropriate for small animal patients. Some of these ventilators were designed specifically to support anesthetized veterinary patients, whereas others were designed for human use, but are applicable to veterinary patients. The classification, principles of operation, and other points about the general function of each ventilator are included. Before operating a ventilator, the user should consult the operation manuals and follow all preuse evaluation procedures recommended by the manufacturer.

Drager Small Animal Ventilator (SAV)

This ventilator (Fig. 18.44) was marketed as an optional component for the Drager Narkovet 2 Anesthesia Machine, but was available on a mobile stand (universal pole) specifically designed for the ventilator.[56] Presently, the ventilator is not being manufactured, but these ventilators remain in use for veterinary anesthesia. The SAV is classified as double circuit, tidal volume preset, and time cycled, with an ascending bellows; it is pneumatically powered and has fluidic circuitry. The pressure of the driving gas should be between 40 to 60 pounds per square inch (psi). The controls include a power ("on-off") switch, a tidal volume adjustment rod to set the attached plate within the bellows housing to the selected tidal volume (200 to 1600 mL), a frequency control knob (10 to 30 breaths/min), and an inspiratory flow knob to control the rate of flow into the bellows housing to drive the bellows. The inspiratory flow knob should be set so that the bellows is fully compressed at the end of the inspiratory phase; however, the bellows should not be deformed at the end of inspiration. Deformation of the bellows at the end of inspiration my indicate an increase in tidal volume by as much as 100 mL. The inspiratory flow control setting affects the peak pressure that

Fig. 18.44. Drager SAV (small animal ventilator; front view). The ascending bellows and the bellows housing with the tidal volume marked in milliliters are shown in the *bottom* of the photograph, and the inspiratory-flow control knob (*left*), the frequency control knob (*center right*), and the power switch (*far right*) are shown in the *top* of the photograph.

is achieved on inspiration and the inspiratory time. Higher inspiratory flows produce shorter inspiratory times and tend to produce higher peak inspiratory pressures. The ratio of inspiratory to expiratory time phase is preset to 1:2. This ventilator provides only controlled ventilation. The ventilator relief valve behind the bellows chamber compensates for the continuous entry of fresh gases into the breathing system. Because the ventilator uses an ascending bellows, the effect of gravity on the bellows maintains a PEEP of approximately 2 cm H_2O.

Before using the ventilator, the proper connections to the gas-supply and scavenger system should be made, and the appropriate preuse checkout procedures should be done for all equipment. Assuming that the anesthesia machine, breathing system, and ventilator are functional, the following is a reasonable step-by-step approach to the operation of this ventilator with a circle breathing system:

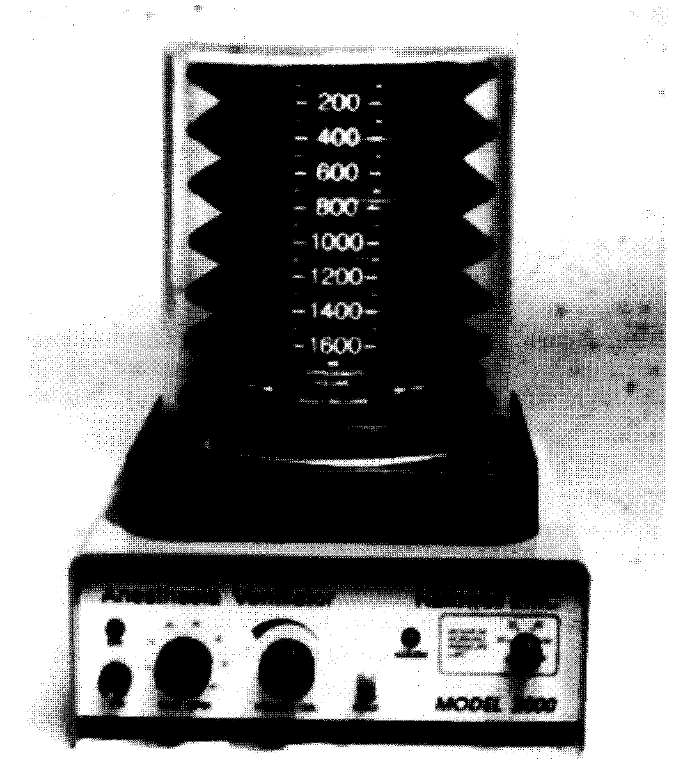


Fig. 18.45. Hallowell EMC Model 2000 Small Animal Veterinary Anesthesia Ventilator (front view). The bellows shown is the medium sized for tidal volumes between 300 and 1600 mL, and the basic control knobs and alarm indicator light are on the front panel. Photograph courtesy of W. Stetson Hallowell.

1. The tidal volume adjustment rod is set appropriately for the patient.
2. Corrugated tubing from the ventilator's breathing-hose terminal is connected to the circle system's reservoir-bag mount.
3. The circle system's APL valve (adjustable pressure-limiting or pop-off valve) is closed.
4. The ventilator's power switch is turned on.
5. The frequency of ventilation is adjusted to approximately the desired number of breaths per minute.
6. The inspiratory flow control knob is adjusted to produce the desired inspiratory time to deliver the preset tidal volume.
7. Frequency of ventilation and inspiratory flow may need to be readjusted to achieve the desired rate of breathing and inspiratory time.

Hallowell EMC Model 2000 Small Animal Veterinary Anesthesia Ventilator

This ventilator (Fig. 18.45) is designed for use with standard small animal anesthesia machines and breathing systems, and the connections to the breathing system, scavenger, and driving gas are shown in Fig. 18.46.[57] This ventilator is classified as double circuit and time cycled with an ascending bellows; it is pneumatically and electrically powered. The ventilator is essentially volume constant within the practical limits described earlier. The ventilator is pneumatically driven and electronically controlled by an electrically activated solenoid valve that allows gas pressure to be supplied to the volume control during the inspiratory phase of the ventilatory cycle. The ventilator's power switch is incorporated into the respiratory-rate control. Therefore, the ventilator is on when the rate selector is turned from the off position to the desired frequency of respiration (6 to 40 breaths/min). The pressure of the driving-gas supply (either oxygen, nitrogen, or clean dry air) should be regulated between 30 to 60 psi. This high flow is necessary only for larger patients.

Fig. 18.46. Hallowell EMC Model 2000 Small Animal Veterinary Anesthesia Ventilator (rear view). Note the connectors on the bellows housing for the breathing system, scavenger system (exhaust), and driving gas. Photograph courtesy of W. Stetson Hallowell.

The control module of the ventilator has the following adjustable components: the on-off and respiratory-rate control knob, a volume control knob, an inspiratory hold pushbutton, and a maximum working pressure limit (MWPL) selector. The ratio of inspiratory to expiratory time phase is preset at 1:2. However, this ventilator is available with an optional adjustable I-E ratio in the range of 1:1.5 to 1:4, enabling users to minimize the inspiratory time when ventilating at a lower frequency of ventilation. The volume control is a variable orifice-metering valve that regulates the driving-gas flow, which compresses the bellows. Basically, the volume control is used to set minute volume. It regulates the inspiratory flow rate directly, and a higher inspiratory flow rate at any given respiratory rate will produce a greater tidal volume. The inspiratory hold pushbutton interrupts the ventilatory cycle and prevents discharge of gas from the bellows housing until the button is released or the MWPL is reached. The MWPL can be set between 10 and 60 cm H_2O. If the MWPL is reached at any time, the inspiratory phase of ventilation is terminated and exhalation is allowed. Low breathing-system pressure will be detected if pressure at the end of inspiration is less than 5 cm H_2O, and a red warning light will illuminate and an alarm will sound indicating the possibility of a disconnection of the patient circuit from the ventilator. This

Fig. 18.47. Hallowell EMC Model 2000 Small Animal Veterinary Anesthesia Ventilator (front view). The bellows shown is the largest one for tidal volumes between 0 and 3000 mL. The basic control knobs and alarm indicator light are on the front panel. Photograph courtesy of W. Stetson Hallowell.

ventilator provides for controlled ventilation; assisted ventilation is not an option.

Three sizes of interchangeable bellows and bellows housings are available to enable various sizes of patients to be ventilated effectively (Fig. 18.47). With the proper bellows, the manufacturer indicates that tidal volumes as small as 20 mL and as large as 3 L can be delivered and that the patient can effectively breathe spontaneously from the bellows when the ventilator is not in operation. The ventilator relief valve compensates for the continuous entry of fresh gas into the breathing system, and the resistance of the relief valve creates a PEEP of 2 to 3 cm H_2O.

Before using the ventilator, connections to the gas-supply and scavenger system should be made, and the appropriate preuse checkout procedures should be done. Assuming proper function of the anesthesia machine, breathing system, and ventilator, the following is a reasonable operational approach for this ventilator with a circle breathing system:

1. The MWPL selector (Fig. 18.45) is set to the desired maximum pressure (safety limit), and the pressure transducer (Fig. 18.46) is connected to the breathing system according to the manufacturer's recommendations.
2. Corrugated tubing from the ventilator's breathing system connector is attached to the circle system's reservoir-bag mount, and the ventilator is attached to the scavenger system.
3. The circle system's pop-off (APL) valve is closed.
4. The ventilator's volume control is adjusted to the minimum setting.
5. The ventilator's power-rate switch is turned on, and the desired frequency of ventilation is set.
6. The volume control knob is adjusted to produce a flow of gas during inspiration that produces the desired tidal volume and/or peak inspiratory pressure.
7. During maintenance, the minute volume is adjusted with the volume control, and the rate control can be used to adjust the size of each tidal volume.

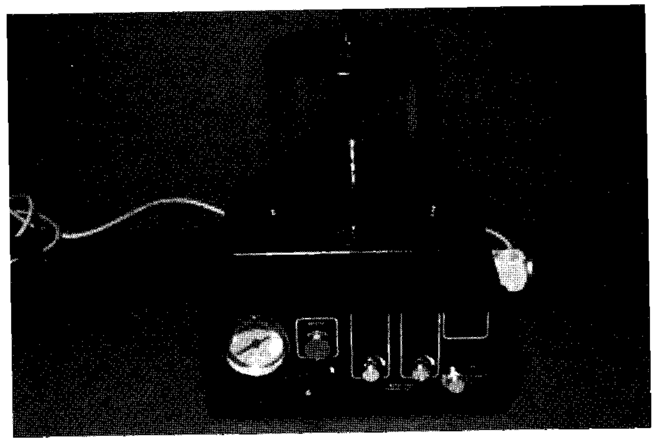


Fig. 18.48. Mallard 2400V Small Animal Anesthesia Ventilator. The bellows is collapsed on the floor of the bellows housing. The tidal volume control is set at approximately 1600 mL. The control knobs on the console are described in the text.

Mallard Medical Model 2400V Anesthesia Ventilator

This ventilator[58] (Fig. 18.48) was originally designed to allow continuous mechanical ventilation of anesthetized pediatric and adult human patients. It is sold to veterinarians as a stand-alone unit for use with breathing system and anesthesia machine. Classified as a double-circuit ventilator, it has electric and pneumatic power sources. The ventilator is controlled by a microprocessor, and the manufacturer describes the ventilator as electronically time cycled and volume limited. The tidal volume is selected by limiting the upward expansion of the bellows. Tidal volume is adjusted by moving a cylinder within the bellows housing to coincide with the desired setting in milliliters, and the cylinder within the bellows housing is secured by a control knob (nut) located on the top center of the housing. This ventilator employs an ascending bellows. The bellows is pneumatically driven, and the ventilator operates at a pressure of 50 ± 10 psi.

The controls are positioned on a console, which is located below the bellows housing. A master on/standby/off switch is present in the right lower corner of the console's front panel; the standby mode allows preselection of respiratory rate and inspiratory time, and the I-E ratio is computed and displayed digitally on light-emitting diode (LED) displays before mechanical ventilation is initiated. Respiratory rate and inspiratory time are controlled by ten-turn potentiometers to allow selection of 2 to 80 breaths/min (respiratory rate) and 0.1 to 3.0 s (inspiratory time), respectively. The I-E ratio display shows the relationship of inspiratory time to expiratory time, giving inspiratory time a value of 1. A black control knob located in the lower left portion of the front panel allows adjustment of inspiratory flow rate (10 to 100 L/min), and a display gauge near the control knob indicates whether the flow being used is low, medium, or high. A green pushbutton is located in the front center portion of the control console; this button activates inspiration as long as the button is pushed in. This button can be used to maintain mechanical ventilation in the event of a power failure and can be used to sigh the patient.

Two sizes of bellows are available. The adult bellows provides tidal volumes ranging from 200 to 2200 mL; the pediatric bellows produces volumes ranging from 50 to 300 mL. An exhalation valve assembly is located on the back of the control console. This valve is closed pneumatically during the inspiratory phase of ventilation, and it opens automatically during the expiratory phase. Excess gas from the patient circuit exits through this valve to prevent the buildup of excessive pressure. The post (19 mm) of this valve should be attached to a scavenger system for elimination of waste gases from the working environment. With an ascending bellows, PEEP (usually 2 or 3 cm H_2O) will be present. In addition, PEEP of up to 20 cm H_2O can be added to the system with the control knob of the optional PEEP valve. Also, an adjustable overpressure relief valve within the console is preset to 80 cm H_2O, and this limits the maximum pressure that can be developed in the patient breathing circuit. Externally, this pressure can be adjusted from 20 to 100 cm H_2O. This ventilator has audible alarms if the ventilator fails to cycle or if an electric power failure occurs. In addition, the LED displays will indicate selection of an inverse I-E ratio, failure of the ventilator to cycle, and low supply-gas pressure (<30 psi).

Before using the ventilator, the proper connections to the gas-supply and scavenger system should be made, and the appropriate preuse checkout procedures should be done for all equipment. Assuming proper function of the anesthesia machine, breathing system, and ventilator, the following is a reasonable operational approach for this ventilator with a circle breathing system:

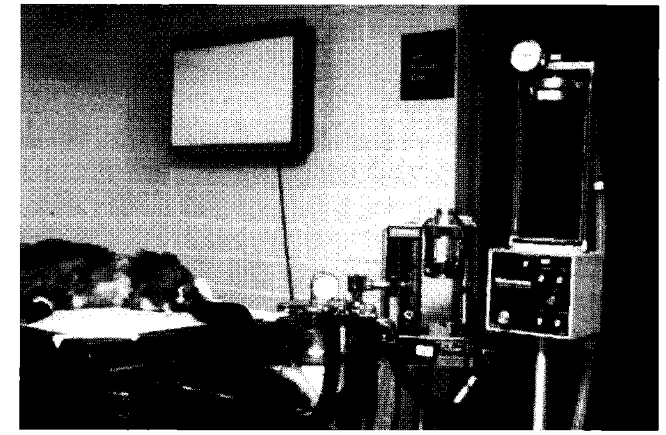


Fig. 18.49. Metomatic Veterinary Ventilator. The ventilator's bellows is connected by a corrugated breathing tube to the reservoir-bag port of the circle breathing system to enable controlled ventilation. Anesthesia was maintained with halothane in oxygen in this dog.

Fig. 18.50. Control panel of a Metomatic Veterinary Ventilator. The function of the various controls of this ventilator are discussed in the text.

1. Prior to clinical applications, refer to the operation manual for instructions and conduct performance verification procedures.
2. Select the appropriate control settings for the tidal volume by limiting the upward expansion of the bellows.
3. Place the master switch in the *standby* mode and dial the desired settings for the respiratory rate and the inspiratory time, based on the patient's needs.
4. Set the inspiratory flow control to the desired rate of flow—low, medium, or high—depending on the needs of the patient.
5. Connect the corrugated tubing from the ventilator's bellows to the circle system's reservoir-bag mount and attach the ventilator to the scavenger system.
6. Close the circle system's pop-off (APL) valve.
7. Set the master switch to the *on* position.
8. The ventilator should cycle according to the selected settings, and only minor adjustments should be necessary (i.e., slight alterations in inspiratory time).

Metomatic Veterinary Ventilator

This ventilator is shown in Figs. 18.49 and 18.50. This unit was designed to ventilate anesthetized small animals being maintained with circle breathing systems. The ventilator is no longer being manufactured, but many units are still in operation in veterinary hospitals.

This ventilator is classified as double circuit and time cycled, with fluidic circuitry and a descending bellows. Within the limits of the definitions, it can be used as a volume-preset ventilator or as a pressure-limited ventilator. The ventilator is powered pneumatically and will function properly with an oxygen-supply pressure to 45 to 55 psi.

Controls (Fig. 18.50) for this ventilator are as follows:[59,60] power (on-off) switch, tidal volume control, inspiratory flow-rate control, expiratory time control, expiratory flow-rate control, inspiratory hold pushbutton, and inspiratory trigger-effort control. The power switch controls a valve that supplies pneumatic power (oxygen at 50 psi) to the ventilator. The tidal volume control adjusts the bellows from 0 to 1400 mL. The inspiratory flow-rate control regulates the rate of delivery of gas from the bellows to the patient during inspiration and is adjustable from 20 to 70 L/min. The inspiratory pressure control sets the maximum pressure that can be delivered to the patient circuit during inspiration, up to 40 cm H_2O; pressure is adjustable from 10 to 40 cm H_2O. The expiratory time control adjusts the time between the end of one inspiratory phase of respiration and the beginning of the next and can be varied from less than 1 to at least 12 s; essentially, it is a setting for respiratory rate although rate is influenced to some degree by other controls. The expiratory flow-rate control allows variation in the rate at which the bellows descends to the fully extended position and is adjustable from 15 to 100 L/min. The inspiratory hold pushbutton allows the initiation of inspiration at any point during the respiratory cycle by depressing and immediately releasing the button. If the pushbutton is depressed and held, inspiration will be initiated, and the bellows will remain at the end-inspiratory position until the button is released. The inspiratory trigger-effort control sets the sensitivity of the ventilator to the negative pressure produced by the patient's inspiratory effort. The setting can be low, which would require only a slight negative pressure to initiate a cycle, or high, which prevents the patient from triggering inspiration; the setting is adjustable from -0.5 to -5.0 cm H_2O. Many of these ventilators were equipped with a patient circuit pressure gauge (manometer) mounted on top of the bellows housing. This ventilator can be set to provide controlled or assisted ventilation.

The ventilator provides a relief valve (pop-off valve) to allow the escape of excess gases that are delivered to the patient circuit. Generally, the pressure in the patient circuit returns to zero at end expiration, since a descending bellows is employed.

Four modes of ventilation can be employed with this ventilator:

1. *Controller, volume controlled and pressure limited.* The rate and pattern of respiration are controlled by the ventilator. The selected tidal volume is delivered as long as inspiratory pressure does not exceed 40 cm H_2O. If the pressure limit is reached before the entire tidal volume is delivered, inspiration will cease.

2. *Controller, pressure controlled and volume limited.* The rate of ventilation is controlled by the ventilator. The ventilator delivers gas to the patient until the preset pressure limit is reached or until the contents of the bellows are fully discharged. Since tidal volume is affected by pressure, changes in airway resistance and compliance of the lungs can alter tidal volume.

3. *Assistor-controller, volume controlled and pressure limited.* The respiratory cycle is initiated by any spontaneous inspiratory effort on the part of the patient. The minimum frequency of ventilation is set by the ventilator, and if the patient fails to initiate the preset number of breaths, the ventilator will cycle at the minimum frequency. The preset tidal volume is delivered unless the inspiratory pressure reaches 40 cm H_2O, at which point inspiration will cease.

4. *Assistor-controller, pressure controlled and volume limited.* The respiratory cycle is initiated by spontaneous inspiratory efforts, and the minimum frequency is set by the ventilator. The patient may initiate a faster rate of respiration. The ventilator delivers gas to the patient until a preset pressure is reached or until the bellows is fully discharged. The tidal volume will be affected significantly by changes in compliance of the lung and airway resistance.

When using the Metomatic ventilator, the first mode (controller, volume controlled and pressure limited) is most frequently used. Before using the ventilator, the proper connections to the gas-supply and scavenger system should be made, and the appropriate preuse checkout procedures should be done. Assuming that the anesthesia machine, breathing system, and ventilator are functional, the following is a step-by-step approach to the operation of the ventilator with a circle breathing system:

1. Select the desired tidal volume.
2. Set the inspiratory trigger-effort control to a high setting, but not to the maximum.
3. Turn the inspiratory pressure control to a high setting (the maximum setting or high enough to assure that the bellows will deliver a complete tidal volume).
4. Set the inspiratory flow-rate control to a midrange setting. After the ventilator is in use, this control will be reset to deliver the tidal volume in approximately 1 to 1.5 s.
5. Set the expiratory flow rate to a midrange setting. This setting can be refined after the ventilator is in use; a setting is usually employed that will not impede ventilation.
6. Set the expiratory time control to a midrange setting. This control should be reset to allow the appropriate frequency of ventilation after the ventilator is in use.
7. Connect the corrugated tube from the ventilator's bellows to the circle system's reservoir-bag port.
8. Close the pop-off (APL) valve of the circle.
9. Turn the power switch on.
10. Observe the character and rate of ventilation, and refine the adjustments of the various controls. Usually, inspiratory flow rate is adjusted first, followed by frequency or expiratory time, and expiratory flow rate.

Fig. 18.51. Ohmeda 7000 Ventilator. The control module is shown with six controls on the *left* two-thirds of the panel and six warning indicators on the *right* two-thirds of the panel. This ventilator is equipped with a pediatric bellows assembly (0 to 300 mL for tidal volume).

Ohmeda 7000 Electronic Anesthesia Ventilator

The Ohmeda 7000 (Fig. 18.51) is a double-circuit ventilator with a pneumatically driven ascending bellows. The ventilator is electronically controlled with a preset minute volume. It is specifically designed as an anesthesia ventilator and can be fitted with either an adult or a pediatric bellows, and its application in human anesthesia has been described.[50,54,61] This ventilator and upgraded models are available for use in human patients[54] and are readily applicable to small animal anesthesia. The control module (Fig. 18.51) has six controls, including the minute volume dial (2 to 30 L/min with the adult bellows and 2 to 12 L/min with the pediatric bellows), the respiratory-rate dial (6 to 40 breaths/min), the I-E ratio dial (1:1, 1:2, and 1:3), power (on-off) switch, the sigh switch (to provide a "sigh" equal to 150% of the tidal volume once every 64 breaths), and a manual cycle button (used to manually initiate a ventilatory cycle only during the expiratory phase). The scale on the bellows housing ranges from 100 to 1600 mL on the adult bellows and from 0 to 300 mL on the pediatric bellows. The bellows assembly exhaust port is 19 mm OD, the connection to the anesthesia machine is 22 mm, and there is a high-pressure (50 psi) diameter index safety system (DISS) fitting for an oxygen line for the driving-gas circuit. The control module of the ventilator computes tidal volume, inspiratory time, expiratory time, and inspiratory flow based on the settings of the various control dials. The bellows should be fully distended (starts at the zero mark on the bellows housing scale) before inspiration begins.

The driving-gas supply is oxygen at 50 psi, which is reduced to 38 psi by a precision regulator within the ventilator; the gas line with the reduced pressure connects to a manifold of five solenoids. Electronic controls regulate the solenoid valves to deliver flows in 2-L/min increments from 4 to 60 L/min. Based on control settings, a precise volume of gas (equal to the tidal volume) is delivered to the bellows chamber to drive the bellows during the inspiratory phase, which forces gas from the bellows into the patient circuit. Flow stops when the full tidal volume has been delivered. A high-pressure relief valve opens at a pressure of 65 cm H_2O if such pressures should occur. During the expiratory phase, gas from the patient circuit (flow from the anesthesia machine) enters the bellows. The ventilator relief opens when the bellows is fully distended and a pressure of 2.5 cm H_2O has been exceeded; excess gas from the patient circuit is vented into the scavenger system.

The manufacturer recommends a bellows assembly-leak test. With the ventilator attached to a circle breathing system with the breathing system's pop-off valve closed, the Y piece occluded, all fresh gas flow off, and the bellows filled from the anesthesia machine's oxygen-flush valve, the bellows should drop no more than 100 mL/min. If a significant leak is present, the ventilator should not be used until the leak is sealed. If the anesthesia machine, breathing system, and ventilator are all in proper working order as indicated by preuse checkout procedures, the following guidelines are appropriate for use of the ventilator:

1. Properly connect the electric and pneumatic power sources for the ventilator.
2. Using the control dials, set the desired values for minute volume, respiratory rate (frequency), and I-E ratio.
3. Make the appropriate connections from the ventilator bellows to the circle system's reservoir-bag port and to the scavenger system.
4. Close the pop-off (APL) valve of the circle system.
5. Be sure that the bellows is completely filled with oxygen-anesthetic mixture.
6. Switch the power control to *on*.
7. Make final adjustments in minute volume and respiratory rate to meet the needs of the patient.

The next generation of ventilators from Ohmeda is the 7800 series of ventilators. The 7800 is available as a stand-alone unit and potentially could be applied to small animal patients. The ventilator is classified as electronically controlled, pneumatically driven, and tidal volume preset, and can accurately deliver tidal volumes of 50 to 1500 mL. A major difference between the 7000 and 7800 series is that tidal volume, rather than minute ventilation, is selected by the operator, which appears to be a significant advantage.[61]

Vet-Tec Small Animal Ventilator (SAV-75)

This ventilator is designed for use in small animal anesthesia.[62] The bellows is designed to ascend on expiration and is pneumatically driven by a Bird ventilator. Used without a bellows, Bird ventilators are classified as single-circuit ventilators, but the SAV-75 performs as a double-circuit unit. This ventilator can be used for ventilation in assist, control, or assist-control modes. When the system is operating, the Bird ventilator supplies gas to pressurize the space between the bellows and the bellows housing (canister) to force the bellows downward, which delivers gases from the bellows through the interface hose (corrugated breathing tube) to the breathing system. The controls on the Bird ventilator include inspiratory pressure, inspiratory flow rate, expiratory time (apnea control), and inspiratory sensitivity. In addition, a manometer, a hand timer (push-pull mechanism), and a DISS connector for the source of pneumatic power are prominent features of the ventilator. Inspiratory pressure can be varied to a maximum of 60 cm H_2O, inspiratory sensitivity from -0.5 to -5.0 cm H_2O, expiratory time to produce 4 to 60 controlled breaths/min, and inspiratory flow to a maximum of 70 L/min. Safety relief occurs on inspiration if a pressure of 65 cm H_2O develops. The pneumatic power source should be delivered to the ventilator inlet at 50 psi. The bellows can deliver a tidal volume of up to 2000 mL. Inspiration can be started or stopped by use of the hand timer. The Bird ventilator is time cycled unless the push-pull manual cycling rod is pulled out, causing the ventilator to be pressure cycled.[55] Figure 18.52 shows a Bird ventilator.

Before using the SAV-75 ventilator for controlling ventilation during anesthesia, the power-supply and scavenger system should be connected, and the appropriate preuse checkout procedures should be done for all equipment. Assuming proper function of the anesthesia machine, breathing system, and ventilator, the following is a reasonable operational approach for the ventilator with a circle breathing system in the control mode:

1. Set the inspiratory sensitivity control to a high setting to eliminate the possibility of patient-initiated ventilation.
2. Set the inspiratory pressure control to the range of 15 to 20 cm H_2O and readjust the setting to achieve the desired tidal volume after steps 5 and 6 have been completed.
3. Connect the corrugated hose (interface hose) from the ventilator's bellows to the reservoir-bag port of the circle system.
4. Close the pop-off (APL) valve of the circle system. At this point, the bellows may need to be filled by increasing the flow of oxygen to the patient circuit (oxygen flowmeter of the anesthesia machine).
5. Turn the inspiratory flow control on to start the ventilator and set the flow control to deliver a tidal volume in approximately 1 to 1.5 s.