Rabbits and rodents are popular pets in urban society because they require little space, are relatively easy to care for, and are a species with which the client can develop a bond. Rodents are generally short lived with the exception of hystricomorphs (guinea pigs, chinchillas, etc.). They have evolved to be prolific to compensate for their short life span. A variety of medical conditions are best treated surgically. Before surgery can be performed, the patient must be anesthetized. The greatest challenges of anesthesia and surgery of pet rodents and rabbits stem from their small size. In recent years, drugs, equipment and techniques have been made available which allow the veterinarian to apply standard anesthetic techniques rabbits and rodents.

**Preanesthetic Evaluation** - Careful and critical preanesthetic evaluation of the small rodent is essential for a successful outcome. A thorough history, physical examination and laboratory data base will provide the anesthesiologist with important information regarding the health status of the patient. Ideally, a preoperative CBC, chemistry profile and complete urinalysis are evaluated and easily obtained in rabbits. In rodents, it may be difficult to get a blood sample due to size but many blood analysis machines can provide a lot of information with a very small sample of blood. Minimally, the PCV, total protein, BUN, blood glucose and urine specific gravity should be determined. Rodents urinate frequently, do not store large amounts in the bladder, and void when stressed making it difficult to get a sample by cystocentesis. Yet, renal insufficiency is common in older rodents, especially hamsters. A reasonable sample can be obtained by placing the patient in a clean glass or plastic container. Watch the patient closely and as soon as it urinates, remove the patient and collect the sample. Dilute urine with high protein is suggestive of renal insufficiency.

An accurate body weight must be obtained prior to anesthesia to be able to calculate drug doses. The response to anesthetic agents is highly variable in small rodents and age, sex, weight and time of the day can affect the patient's response to anesthetic drugs. Before anesthesia, it is best to calculate doses and draw up emergency drugs. Epinephrine, atropine, and lidocaine are the most commonly used drugs during cardiopulmonary arrest and are inexpensive. Being prepared will avoid panic during an emergency and having to find and calculate emergency drug doses.

Prior to anesthesia, any underlying diseases, especially respiratory disease commonly seen in rabbits and rodents should be treated. Stress can induce respiratory disease in subclinical patients. Perioperative antibiotics can help keep subclinical respiratory infections from becoming a life threatening problem. If the animal is dehydrated, administration of fluids prior to anesthesia is a priority. Fluids are best administered IV or IOs in critically ill patients, but subcutaneous fluids are appropriate in many situations. Crystalloid fluids are administered during anesthesia at the standard rate of 10 ml/kg/hr. If the procedure is expected to take more than 20-30 min. the 5%dextrose in water should be used. Warming the fluids is helpful in minimizing hypothermia.

Since rabbits and rodents cannot vomit, pre-anesthetic fasting is not required. A short fast is beneficial in allowing the patient to swallow any food within the oral cavity minimizing the risk of aspiration or of food being carried into the trachea during intubation. A fast of 1-2 hrs is recommended prior to dental procedures.

**Preanesthetic Medications** – Administration of preanesthetic medications is routine in rabbits and rodents. Parasympatholytics will decrease vagal tone as well as the amount of bronchial and salivary secretions. Atropine (0.05 mg/kg SQ; 0.08 mg/kg SQ in rabbits) or glycopyrrolate (0.01 to 0.02 mg/kg SQ or IM; 0.01-0.1 in rabbits) can be used. Atropine has a more rapid onset and is recommended for treating bradycardia and cardiac emergencies while glycopyrrolate is more slowly absorbed but has a longer duration. Up to 50% of rabbits have plasma atropinesterase and the dose is higher and it must be given more frequently. As a preanesthetic, glycopyrrolate is preferred in rabbits. These drugs can thicken respiratory secretions which can obstruct the airway.
Preemptive analgesia is recommended in patients undergoing surgery. Buprenorphine (0.01-0.05 mg/kg SQ) given 20-30 min. prior to induction will provide some sedation and preemptive analgesia. The dose of butorphanol varies widely across rodent species but is also a useful pre-anesthetic opioid (see table in Flecknell). A dose of 0.1-0.5 mg/kg IM or IV is recommended in rabbits. While nonsteroidal anti-inflammatory drugs are useful analgesics, they are best used postoperatively because they can have serious negative effects on renal function if the patient's blood pressure drops during anesthesia.

Preanesthetic administration of sedative tranquillizers is often beneficial in rabbits in rodents to help minimize stress and anxiety, and to minimize patient struggling. The benzodiazepines are known for their anxiolytic properties and midazolam is very useful in calming rabbits and rodents (0.5-5 in rabbits and 3-5 mg/kg in rodents IM or IV). It is also reversible with flumazenil (0.1 mg/kg IV – may precipitate seizure).

Ketamine HCl either alone or in combination with diazepam, medetomidine or acepromazine may be given at the lower end of the dosage will allow faster induction of anesthesia with an inhalation agent and minimize stress. It also lowers the maintenance requirements of the inhalation agent.

Injectable Agents - A variety of injectable agents have been used either alone or in combinations to induce and/or maintain anesthesia. Depending on the drug and the dose being used, light sedation or a surgical plane of anesthesia can be achieved. Most commonly, the dissociative anesthetic ketamine HCl either alone or combination with acepromazine, medetomidine, diazepam, midazolam, and xylazine is used (see table in Cantwell). It has been suggested that acepromazine should not be given to gerbils because they are prone to epilepsy; however, there is no scientific evidence that acepromazine lowers the seizure threshold. The benzodiazepines (diazepam and midazolam) can be reversed with flumazenil and the alpha 2 agonists (medetomidine and xylazine) can be reversed with atipamazole or yohimbine which is helpful during an emergency or if recovery is prolonged.

Ketamine alone produces poor muscle relaxation, little or no analgesia, muscle irritation at the injection site and respiratory depression. Depending on the dose being used, it will produce adequate restraint and sedation for minor procedures. A combination of ketamine with one of the above mentioned drugs will greatly improve muscle relaxation and smooth the anesthetic experience.

Tiletamine/Zolazepam (Telazol®) has been used with good success in small rodents. A dose of 6-10 mg/kg IM is adequate prior to inhalation anesthesia. Its advantages over ketamine include better muscle relaxation, small volume of injection, rapid induction time and a wide margin of safety. Disadvantages of this drug combination are increased respiratory secretions, variability in recovery times, and short self life after reconstitution.

If venous access is established, propofol (10-25 mg/kg) provides rapid induction and recovery, but it is only effective if administered IV. It can also be used for maintenance of anesthesia if a short procedure is being performed. Fentanyl-droperidol should not be given to guinea pigs IM since it may cause severe muscle necrosis.

Some examples of injectable anesthesia protocols that have been used with good results are:

**Rodents**

1. Midazolam (5 mg/kg) + ketamine (100 mg/kg) + buprenorphine (0.05 mg/kg) IP
2. Acepromazine (1 mg/kg) + ketamine (100 mg/kg) IP
3. Xylazine (5 mg/kg) + ketamine (50 mg/kg) + atropine (0.05 mg/kg) IP

**Rabbits**

1. Midazolam (0.05 mg/kg) + buprenorphine (0.03 mg/kg) + ketamine (10 mg/kg) IM
2. Diazepam (1.0 mg/kg) + ketamine (10 mg/kg) IM
3. Acepromazine (0.2 mg/kg) + oxymorphone (0.1 mg/kg) + glycopyrrolate
**Inhalation Agents** - Building the anesthetic regimen for rodents on the administration of inhalation agents is preferred as inhalation agents allow better control of anesthetic depth, are safer, provide for the administration of oxygen and, if the animal is intubated, allow for control of the airway. Induction of anesthesia can be achieved by placing the animal in an induction chamber. For small patients chambers are easily made from plastic containers or glass tanks with appropriate tops. Induction with inhalation agents via face mask is preferred, since this will allow for closer control of the animal and intervention in the event of complications but restraint often causes an unacceptable level of stress. The smallest mask possible should be chosen to minimize dead space.

Endotracheal intubation is difficult in rodents due to their small size and the inability to open their mouth wide enough to visualize the glottis. As a result, inhalation anesthesia is often maintained using a face mask. For thoracic surgery intubation is essential and for oral/dental procedures it is ideal. While it takes practice and experience to perform endotracheal intubation, it can be accomplished. In guinea pigs and hamsters which often store food in their mouth or cheek pouches, removal of food from the oral cavity is recommended before attempting to intubate the animal. Proper equipment should be available and the inexperienced should not spend excessive amounts of time attempting to intubate the animal, as it is critical to keep the anesthetic event as short as possible. The most complications that occur following repeated unsuccessful attempts at intubation are related to trauma to the larynx and pharynx.

A small endoscope (0.9-1.0 mm) can serve as a stylet within the endotracheal tube and allow visualization of the glottis. Advance the endotracheal tube (available in sizes as small as 1 mm) listening for respiratory sounds. Place 0.1 cc of 2% lidocaine into the tube and blow to spray it onto the glottis to prevent laryngospasm. Wait 5 min. for it to take effect. Place the endoscope inside the endotracheal tube and advance the scope into the trachea. Slide the tube off the scope and remove the endoscope. Confirm tube placement (move bag, end-tidal CO₂, tube fogging) and secure the tube in place. Because the tube is easily dislodged during patient positioning, etc. consider placing butterfly tape around the tube at the mouth and suturing it in place. In small species an intravenous catheter can be used as an endotracheal tube.

An alternative technique is blind intubation which works well in rabbits and some rodents. There are 3 key hints to success with blind intubation. First, lidocaine is used as described above to minimize laryngospasms. Be sure to wait a full 5 min. to allow it to work. Tip the head way back so that the path from the front of the mouth through the mouth and into the trachea is a straight short. Advance the tube to the level of the glottis. At this location you should hear loud, clear breathing sounds. Erratic, muffled sounds usually indicate the tube is heading down the esophagus. Listen to the rhythm and get in tune with the in, out, in, out... On inspiration, slip the tube into the trachea. Because of the lidocaine, the patient may not cough or react as the tube is inserted. Make sure to confirm the tube is in the trachea as described above.

Isoflurane and sevoflurane are commonly used inhalant agents. Sevoflurane is associated with a more rapid induction and less systolic blood pressure depression. A nonrebreathing system should be used to minimize dead space and resistance to air movement. An oxygen flow rate of 1 l/min is adequate for rodents on a nonrebreathing system.

**Monitoring** - In recent years a variety of monitoring devices have been made available which are practical to use in small patients. It is better to keep the animal at a light plane of anesthesia than allowing it to get too deep. Respiratory depression occurs quickly and, if unnoticed, can result in serious complications. Monitoring the rate and pattern of respiration, cardiac activity, mucous membrane color, and the presence or absence of a variety of reflexes are used to monitor the level of anesthesia. At a surgical plane of anesthesia, the palpebral reflex should be absent, the corneal reflex present, the respiratory pattern deep and regular, and the toe pinch (pedal) reflexes absent. In guinea pigs the pedal reflex may still be present at a surgical plane of anesthesia. This should not be confused with the patient being too light. Loss of corneal reflex and presence of dilated pupils are signs of deep anesthesia and cerebral hypoxia; however, ketamine and parasympatholytics also cause pupil dilation.

A Doppler flow probe positioned at the ventral tail base or on the heart will indicate heart rate and flow of blood (not just electrical activity). An electrocardiograph monitor placed in the conventional manner will also provide continuous monitoring of electrical cardiac activity but must be capable of reading high heart rates (300 in
rabbits, 600 in mice). Pulse oximeters to measure hemoglobin saturation with oxygen are useful and a variety of probes that can be placed at suitable sites is available. Respiration depth and character are best monitored by a support person as end-tidal CO$_2$ monitors and respiratory monitors add an unacceptable amount of dead space to the system.

Indirect blood pressure monitors can be useful in larger patients but the cuffs are often too small for most rodents.

Rabbits and rodents have a high body surface:volume ratio so they rapidly lose body heat when under general anesthesia. As the body temperature decreases, the anesthetic requirement decreases so patients become deeply anesthetized. Eventually, prolonged recovery, bradycardia, and fatal fibrillation occur. Measures for thermal support include circulating warm water blankets, forced warm air blankets, radiant heat lamps, and other devices. Plastic surgical drapes can hold heat in around the patient. Rabbits and rodents lose more heat through their extremities, including their tail, so remember to keep their tails and toes warm!

The eyes of rabbits and rodents are more prominent and frequent lubrication is vital. Under ketamine, the eyelids do not close so it is especially important if ketamine is used. Rays may develop ocular opacity that should resolve quickly postoperatively.

**Recovery** - During the recovery period energy loss should be minimized. Hypothermia decreases the patient's metabolic rate and the rate of excretion of drugs prolonging recovery. Wet hair should be dried and the patient placed on a warm towel or blanket. The patient should be placed in a well ventilated, oxygen enriched, warm environment. Incubators are ideal providing proper temperature control, administration of oxygen, and adequate humidity. If an incubator is not available, a circulating warm water heating blanket, heat lamp and warm water bottles can be used. Patient activity is low during recovery so the ambient temperature should be 20-24 C for the first 24 hr.

Most postoperative patients on analgesics do not take in adequate amounts of fluids. It is important to provide maintenance fluids for at least the first 24 hr. Minimize stress and handling. Administer postoperative analgesics.

**Analgesia** - Providing postoperative analgesia is important in rodent anesthesia (Flecknell). A variety of agents has been used with good success and appears to be effective in providing the recovering patient with analgesia. Buprenorphine (0.05 (rats and g. pigs)-0.1 (hamsters, gerbils, mice) mg/kg SQ q 6-12 hrs is commonly used for postoperative analgesia. Butorphanol can be given postoperatively at a dose of 1-2 (mice, rats, g. pigs) mg/kg SQ q 4 hr. Morphine is not suitable as an analgesic agent in hamsters since this species shows high resistance to its effects but buprenorphine is effective. Nonsteroidal anti-inflammatory drugs including flunixin, ketoprofen and meloxicam have used in rodents with meloxicam (1-2 mg/kg SQ or PO) currently most commonly recommended.

The following resources provide valuable information along with tables with doses for different small mammal species.
