Anesthesia Monitoring
By Darci Palmer, B.S, L.V.T., VTS (Anesthesia)
Tina Branham, L.V.T., VTS (Anesthesia)

I. Importance of Monitoring
A. Gather information about physiologic state of the patient
B. Aid in making decisions about management of anesthesia (depth, fluid therapy, intra-operative pain management, etc)
C. Detect changes that could indicate a detrimental situation and allow for early intervention
D. Legal document detailing events that occur while patient is anesthetized
   - Parameters should be monitored and recorded every 5 minutes
   - Use trend monitoring to interpret the parameters
     ➢ Method used to graph parameters monitored during anesthesia. A line graph shows a trend of highs and lows and can give an idea of baseline values.
     ➢ Monitoring starts before any drugs are administered (obtain a complete history and physical exam of patient) and continues into the recovery period.

II. Monitoring the Central Nervous System
A. Depth of Anesthesia
   - Depth of anesthesia can be determined by evaluating several different reflexes and signs
   - Reflex responses vary between species, individual animals and the drugs administered during pre-medication, induction and maintenance.
   - With most reflexes, loss of reflex indicates an increase in anesthetic depth and a return of a reflex indicates arousal from anesthesia
B. Common reflexes used to judge depth of anesthesia
   1. Eye position
      ➢ Begins in central position, as the patient moves into a light to moderate depth the eye rotates rostroventrally, as the patient moves to a deeper plane of anesthesia the eye rotates back to the central position
      ➢ Gravity has some effect on eye position when the patient is placed in dorsal recumbancy
      ➢ Reliable indicator in the dog and cow
   2. Eye reflexes
      ➢ Palpebral reflex is tested by gently tapping at the lateral or medial canthus of the eye or gently stroking the eyelashes and observing a partial or complete closure of the eyelids (blink).
         ➢ The palpebral reflex is weak to absent at a plane of anesthesia satisfactory for surgical stimulus
         ➢ Can become desensitized if tested to frequently
      ➢ Corneal reflex is tested by touching the cornea with a sterile object (drop of water or artificial tear solution) and noting the blink response
Controversy over use of this reflex

- **Nystagmus** is an involuntary rapid movement of the eyeball usually from side to side (medial to lateral)
  - Presence of nystagmus in horses indicates a light plane of anesthesia
  - Commonly seen in horses at induction and recovery

3. **Pupil constriction/dilation**
- Pupil in an awake patient will constrict and dilate in response to light
- As the patient moves into stage II the pupil becomes slightly dilated in response to excitement
- The pupil returns to normal once the patient reaches stage III. Constriction may be seen as the patient advances through the planes of stage III.
- As anesthesia moves into deeper planes the pupil becomes more and more dilated

4. **Lacrimation**
- Tears are present in an awake animal to keep the eye moist
- Once the patient is anesthetized to an adequate surgical plane lacrimation decreases. It is very important to keep the eyes lubricated during anesthesia
- Lacrimation during anesthesia can be viewed as a sign of light stage anesthesia. Commonly seen in horses.

5. **Swallowing reflex**
- Occurs spontaneously in awake animals, observed by watching ventral neck region
- Reflex is lost at medium depth of anesthesia and regained just before patient recovers consciousness
- During recovery, the swallowing reflex should be present before patient is extubated to prevent aspiration into the lungs

6. **Laryngeal reflex**
- Stimulated when the larynx is touched by an object
- Response is immediate closure of the epiglottis and vocal cords
- If patient is light at induction the laryngeal reflex may be seen making it difficult for intubation
- A sustained laryngeal reflex response is the cause of laryngospasm commonly seen in cats

7. **Pedal reflex**
- Tested by squeezing or pinching a digit or pad and observing if the patient withdraws the limb. This indicates inadequate depth for surgical stimulus.
- Commonly used with injectable anesthetics.
8. **Ear flick reflex or Whisker reflex**
   - Tested by gently touching the hairs on the inner surface of the pinna and observing a twitch of the ear or whiskers in a cat
   - Can become desensitized if reflex is tested too frequently

9. **Muscle Tone**
   - Skeletal muscles become more relaxed as depth increases and offers little resistance to movement
   - Assessed by testing jaw tone and anal tone. Muscle tone is present in light to medium planes of anesthesia and becomes more flaccid as depth increases
   - Some drugs promote muscle relaxation (diazepam, xylazine) and some increase muscle tone (ketamine, tiletamine).

10. **Response to Surgical Stimulus**
    - Movement in response to surgical stimulus indicates inadequate depth
    - Manipulation of viscera can stimulate the patient if adequate depth is not present
    - Patient may pant as a response to surgical stimulus if adequate depth is not present
    - This reflex should not be used alone to judge depth because pain can also elicit these responses.

III. Monitoring the Cardiovascular System

A. **Heart Rate**
   - Heart rate is a major determinant of cardiac output. Cardiac Output equals heart rate times stroke volume (CO = HR x SV)
   - Always obtain a resting heart rate from the patient prior to administering any anesthetic drugs. Note the rate and rhythm.
   - Most anesthetic drugs have a depressant effect on heart rate and myocardial function. Heart rate will decrease from the resting rate in most anesthetized animals. Drugs such as atropine, glycopyrrolate, ketamine and tiletamine will increase heart rate.
   - Normal Values

<table>
<thead>
<tr>
<th></th>
<th>Dog</th>
<th>Cat</th>
<th>Horse</th>
<th>Cow</th>
</tr>
</thead>
<tbody>
<tr>
<td>Awake</td>
<td>70-100bpm</td>
<td>145-200bpm</td>
<td>30-45bpm</td>
<td>60-90bpm</td>
</tr>
<tr>
<td>Anesthetized</td>
<td>50-180bpm</td>
<td>100-220bpm</td>
<td>28-50bpm</td>
<td>48-90bpm</td>
</tr>
</tbody>
</table>

- Heart rate should be higher in small breeds of dogs and much higher in pediatric patients under 3 months of age
• Ways to assess heart rate
  1. **Palpation of peripheral artery**
     - Note rate and pulse quality
  2. **Auscultation of chest wall**
     - Listening with a stethoscope in conjunction with palpating a pulse will give an indication to any pulse deficits that may be present
  3. **Esophageal Stethoscope**
     - Consists of a tube with a balloon on the end that is passed down the esophagus of an anesthetized animal. The open end is connected to an ordinary stethoscope headpiece. The tube should be advanced down the esophagus until the tip is level with the heart or a heart rate can be heard.
     - Provides information about heart rate and rhythm. Can also obtain respiration rate.
  4. **Electrocardiography (ECG)**
     - ECG is an evaluation of the heart’s electrical activity
     - ECG gives information about cardiac arrhythmias and about myocardial environment (hypoxia, hyperkalemia, acidosis, hypercalcemia).
       - The ECG **does not** give any information about the mechanical function of the heart and should not be relied on for the sole method of monitoring cardiovascular function.
     - The white lead is placed on the right front armpit, the black lead is placed on the left front armpit and the red lead is placed on the left hind flank in small animals and along the jugular groove in large animals.
     - Normal heart rhythm (sinus rhythm) originates in the sinoatrial (SA) node of the right atrium. Each waveform generated gives regional information on depolarization or repolarization and conduction in the heart.
       - **P wave** = atrial depolarization
       - **PR (PQ) interval** reflects time taken for the impulse to conduct from the SA node, through the AV node and to the ventricles
       - **QRS complex** = ventricular depolarization
       - **T wave** = ventricular repolarization
       - **QT interval** indicates the duration of ventricular systole
     - Common arrhythmias that can occur during anesthesia include
       1. Sinus bradycardia
       2. Second degree heart block
       3. Escape beats
       4. Ventricular premature complexes
       5. Ventricular tachycardia
       6. Third degree heart block

B. **Tissue Perfusion**
  1. **Mucous membrane color**
     - Should be pink and moist to the touch
- Pale mucous membranes are a result of poor perfusion and may indicate blood loss or anemia.
- Purple or blue mucous membranes indicate cyanosis.
- Dry, clammy, or sticky mucous membranes can indicate that the animal (particularly the horse) may be dehydrated.

2. **Capillary Refill Time (CRT)**
   - Apply pressure to the gums and count the time it takes for color to return.
   - Normal is less than 1.5 seconds.
   - Prolonged CRT is an indication of vasoconstriction which can indicate poor perfusion.

C. **Blood Pressure**
   - Arterial blood pressure can be described using the following terms:
     1. **Systolic Pressure (SAP)** is produced by the contraction of the ventricles and propels blood through the aorta and major arteries.
        - In awake healthy animals the SAP is 140-160mmHg.
     2. **Diastolic Pressure (DAP)** is the pressure that remains when the heart is in its resting phrase, between contractions. It is the lowest pressure that is exerted throughout the cardiac cycle.
        - In awake healthy animals the DAP is 85-95mmHg.
     3. **Mean Pressure (MAP)** maybe calculated by MAP = DAP + 1/3 (SAP-DAP). MAP establishes an adequate perfusion pressure for the major organs.
        - A MAP of 60mmHg in small animals and 70mmHg in large animals is required to ensure adequate minimal blood flow to the kidneys, brain and liver.
        - During anesthesia, hypotension is defined as below 60mmHg in small animals and 70mmHg in large animals.
          - Negative effects of hypotension include: renal failure, reduced hepatic metabolism in dogs, ventilation-perfusion mismatch, hypoxemia, delayed recovery for anesthesia and neuromuscular complications (myopathy).
        - In awake healthy animals the MAP is 90-110mmHg.

- Arterial blood pressure (ABP) usually decreases from the awake values during anesthesia. Normal values for the **anesthetized** patient include:

<table>
<thead>
<tr>
<th></th>
<th>Dog</th>
<th>Cat</th>
<th>Horse</th>
<th>Cow</th>
</tr>
</thead>
<tbody>
<tr>
<td>Systolic</td>
<td>80-120mmHg</td>
<td>80-150mmHg</td>
<td>100-120mmHg</td>
<td>120-150mmHg</td>
</tr>
<tr>
<td>Diastolic</td>
<td>40-80mmHg</td>
<td>40-80mmHg</td>
<td>50-80mmHg</td>
<td>75-100mmHg</td>
</tr>
<tr>
<td>Mean</td>
<td>60-100mmHg</td>
<td>60-120mmHg</td>
<td>70-100mmHg</td>
<td>90-120mmHg</td>
</tr>
</tbody>
</table>
• Neonates or pediatric patients (less than 3 months) have an immature sympathetic nervous system. They are more dependent on heart rate for cardiac output than adults. A lower ABP is tolerated because they have less contractile tissue than adults do. Changes in cardiac output are mediated by changes in rate rather than contractility.

• Ways to evaluate Blood Pressure

1. **Indirect**
   
a. **Oscillometric Technique (Blood pressure cuff)**
   
   - Non-invasive measure of peripheral blood pressure by mechanically inflating a cuff placed around the extremities in small animals or the tail in large animals.
   
   - The cuff is inflated until complete occlusion of blood flow occurs. As pressure is released from the cuff, a transducer located within the monitor records measurements.
     
     - SAP = first oscillation detected
     - DAP = oscillations decrease rapidly, can be calculated
     - MAP = amplitude of oscillations is maximal, usually a calculated value

   - Proper size of cuff is determined by measuring the width of the cuff to the circumference of the limb. The width of the cuff should be approximately 40% the circumference of the limb. An artery should be palpated and the arrow on the cuff should be placed over the artery.
     
     - If the cuff is too tight or too wide the readings will be erroneously low because the cuff itself will partially occlude the underlying artery
     - If the cuff is too loose or too narrow the readings will be erroneously high because excessive cuff pressure will be required to occlude the underlying artery.

   - Movement greatly affects the results and it is unreliable in small dogs and cats with high heart rates or in patients that are hypotensive.
   
   - Use as a trend-monitoring tool. A pulse should be palpated and changes in the reading should be compared in relation to pulse changes.

b. **Doppler**

- Ultrasonic doppler flow detector is connected to a cord that contains the doppler crystal. The doppler crystal is placed over a peripheral artery and taped in place. An audible sound of the pulsating artery should be heard. An appropriately sized cuff is placed proximal to the doppler crystal. A pressure gauge (sphygmomanometer) is attached to the cuff. The cuff is inflated until the pulse sound stops. Slowly deflate the cuff until the pulse is heard.

- Only measures systolic blood pressure
Doppler’s attached to cats have been shown to read 10-15mmHg lower than actual value. Locations for placement of the doppler crystal include proximal to the metacarpal or metatarsal pad, ventral aspect of tongue, medial aspect of humerus, lateral aspect of hock, medial aspect of thigh, lateral aspect of tibia or ventral aspect of tail.

2. Direct
   a. Placing a catheter in a peripheral artery directly monitors arterial blood pressure.
   b. This is the most accurate measurement of arterial blood pressure.
   c. An electronic pressure transducer is attached to the catheter by extension tubing. A cord from the pressure transducer transmits the signal to a monitor and displays SAP, DAP, MAP and a constant waveform.
   d. If equipment is not available, an aneroid manometer can be attached to the arterial catheter. The only reading obtained is the MAP.
   e. Arteries that are commonly catheterized
      - Dog and Cat: palmar common digital arteries, dorsal pedal artery, femoral artery
      - Horse: facial artery, transverse facial artery, lateral nasal artery, metatarsal arteries
      - Ruminants: facial artery, auricular artery and metatarsal arteries
   f. Disadvantages include:
      - Invasive technique that is best placed in anesthetized patients because flushing the arterial catheter with heparinized saline is painful to the awake animal.
      - It must be placed at the same level as the apex of the heart and zeroed. A transducer that is lower than the heart will give a false high reading and a transducer that is higher than the heart will give a false low.
      - Thrombosis and occlusion of flow to the extremity can occur if taped to tightly.
      - If transducer becomes disconnected from the arterial catheter without notice the patient can essentially bleed out.
      - Equipment needed for set up can be expensive

IV. Monitoring the Respiratory System
   A. The overall goal of the respiratory system is to move oxygen into the lungs and remove carbon dioxide from the lungs
   B. Respiration is depressed to some degree by the anesthetic drugs used in pre-medication, induction and maintenance.
   C. Normal values

<table>
<thead>
<tr>
<th></th>
<th>Dog</th>
<th>Cat</th>
<th>Horse</th>
<th>Cow</th>
</tr>
</thead>
<tbody>
<tr>
<td>Awake</td>
<td>10-20bpm</td>
<td>15-25bpm</td>
<td>8-16bpm</td>
<td>10-16bpm</td>
</tr>
<tr>
<td>Anesthetized</td>
<td>8-14bpm</td>
<td>10-14bpm</td>
<td>6-10bpm</td>
<td>6-10bpm</td>
</tr>
</tbody>
</table>
D. Observing the chest excursion of the anesthetized patient is the simplest way to
determine the respiratory rate. If the patient is covered by surgical drapes, observing
movements of the reservoir bag is also acceptable.

E. Ways to monitor oxygen and carbon dioxide

1. Direct
   a. **Arterial blood gas analysis**
      - Gives an indication of ventilation, oxygenation and acid-base status
      - Most accurate method to determine oxygen and carbon dioxide content for
        an anesthetized patient
      - Obtain an arterial blood sample, using an ISTAT machine and cartridge
        the following information can be obtained:
        - **pH**: indicates whether acidemia or alkalemia is present
        - **PaCO$_2$**: partial pressure of CO$_2$ in arterial blood
        - **PaO$_2$**: partial pressure of O$_2$ in arterial blood
        - **HCO$_3$^-**: bicarbonate
        - **TCO$_2$**: total carbon dioxide present in body
        - **BE**: base excess
        - **SaO$_2$**: oxygen saturation in the arterial blood

   - Normal values for arterial blood in awake animals
     |        | Dog       | Cat       | Horse      |
     |--------|-----------|-----------|------------|
     | PaCO$_2$| 34-40 mmHg| 28-34 mmHg| 36-46 mmHg |
     | PaO$_2$ | 85-100 mmHg| 90-110 mmHg| 90-100 mmHg|

   - CO$_2$ should be the same for awake and anesthetized animals
   - All healthy patients on 100% oxygen during anesthesia should have a
     PaO$_2$ between 400-500 mmHg. The predicted PaO$_2$ is approximately 4-5
     times the percentage of inspired oxygen.

   - Permissive hypercapnia in horses
     - Allowing a higher than normal PaCO$_2$ to be present during anesthesia.
     The normal range for CO$_2$ in a horse under anesthesia is 36-46 mmHg.
     The range for CO$_2$ during permissive hypercapnia is 58-69 mmHg.
     Most if not all horses should be placed on a mechanical ventilator for
     procedures over an hour or if the procedure is in depth. During
     mechanical ventilation venous return and cardiac output are decreased.
     When CO$_2$ reaches what we consider the hypercapnic range it acts like
     a sympathetic stimulus (via catecholamine release) and supports
     cardiovascular function, this in turn will indirectly increase arterial
     blood pressure and will also reduce the anesthetic requirement. Note
     that CO$_2$ over 90 mmHg puts the patient into a hypnotic state in which
     it is difficult to evaluate the depth of anesthesia.

2. Indirect
   a. **Capnograph**
      - Non-invasive measurement of carbon dioxide
- Provides breath to breath numerical values of end tidal carbon dioxide (ETCO2)
- ETCO2 is measured through a gas sampling line and reflects the amount of carbon dioxide exhaled by the patient.
- Displays a waveform
  - Upward slope = expiration
  - Downward slope = inspiration, baseline should read zero
- Under ideal conditions ETCO2 reflects the alveolar concentration of carbon dioxide. It is usually presumed that alveolar and capillary carbon dioxide are equilibrated, therefore ETCO2 can be used to estimate PaCO2.
  - ETCO2 under estimates PaCO2 by 10-15 mmHg

b. **Pulse Oximetry**
- Noninvasive measurement of the percentage of hemoglobin saturated with oxygen in arterial blood, symbol is SpO2
- Works on the principle that hemoglobin absorbs red and infrared light at different wavelengths, depending on whether the hemoglobin is bound to oxygen (infrared) or deoxygenated (red). The pulse oximeter uses two light-emitting diodes that pulse red and infrared light through perfused tissue several hundred times per second. The amount of light absorbed at each wavelength is measured by sensitive photodetectors. The absorption data is expressed as a percentage of oxygenated hemoglobin to total hemoglobin.
- Pulse oximeters display a digital record of pulse rate, with an audible beep and some monitors display the oxygen saturation waveform
- Normal range during anesthesia should be around 95%. The percent of saturation gives an indication of the adequacy both of ventilation and of circulation.
- Oxygen-hemoglobin dissociation curve is a graphic representation of the relationship between the percentage of oxygen saturation of hemoglobin (SaO2 or SpO2) and the partial pressure of oxygen in the arterial blood (PaO2)
- Depending on the probe the pulse oximeter can be placed on the tongue, lip, vulva, prepuce, toe web, fold of flank, pinna of ear, metacarpus, digits and tail. As a general rule the probe should be placed on tissue without hair, that is non-pigmented and fairly thin

V. **Monitoring Body Temperature**
A. In normal awake animals, body temperature is regulated by the central nervous system. If an awake animal is cold, common behavior responses include heat seeking, curling up to minimize body surface, piloerection of hair and shivering.
B. General anesthesia inhibits thermoregulation, vasoconstriction and shivering, thereby decreasing the threshold for cold responses.
C. Majority of heat loss in an anesthetized patient occurs through evaporation. Examples include exposure to cold operating room conditions, use of cold solutions for prepping and abdominal exposure during surgery.
D. Ways to prevent hypothermia during anesthesia
   - Warm operating room and recovery room
   - Warm air blankets
   - Circulating warm water blankets
   - Warmed IV fluids and surgical irrigation fluids
   - Wrap extremities of very small dogs and cats with plastic wrap and vetrap or aluminum foil
   - Warm fluid bags surrounding patient

E. Hyperthermia
   - Hyperthermia developing in dogs and cats is most often caused by either excessive application of heat in an attempt to prevent hypothermia or by a pyrogenic reaction to a bacterial infection, a contaminant in IV fluids or drugs.
   - Other causes of intra-operative hyperthermia are loss of central nervous system temperature regulation, thyrotoxicosis or phaeochromcytoma.
   - Hyperthermia is sometimes manifested by malignant hyperthermia syndrome, which is a life threatening hypermetabolic condition triggered by stress and certain anesthetic agents.
   - Hyperthermia can develop in cats during recovery from anesthesia that were induced with tiletamine-zolazepam or ketamine. Increased temperature is associated with increased muscle activity such as paddling, uncoordinated movements or purposeful movements directed at restraints or bandages.

VI. Miscellaneous
A. Monitoring Urine Volume
   - Urine output depends on the renal blood flow which in turn depends on cardiac output and circulating blood volume, thus it is relatively sensitive indicator of the circulatory state during anesthesia
   - Urine output of less than 0.5-1ml/kg/hour is inadequate
   - Horses with full bladders will most likely have a rough recovery from anesthesia; it is always a good idea to catheterize them for procedures that require anesthesia for longer than one hour.

B. Monitoring Blood Glucose
   - Should always be monitored in pediatric patients, diabetics, patients with hepatic disease, portal systemic shunts, insuloma, septicemia or endotoxemia.
   - Consequences of hypoglycemia are coma, hypotension, or prolonged recovery from anesthesia with depression weakness or even seizures
   - Patients at risk should be given dextrose in some form during fluid therapy

C. Monitoring Post-operative Pain
   - Pain assessment should be continued throughout the entire anesthetic period.
   - Parameters that should be evaluated to assess pain include
     - Respiratory rate and pattern
     - Heart rate
     - Anxiety
     - Vocalization
     - Body position
     - Response to manipulation and palpation
Evaluate what analgesic drugs were given peri-operatively and the time they were administered. Opioid agonists can induce dysphoria in some animals, which can be mistaken as vocalization due to pain.
I. Why these techniques are useful in a clinical setting.
   A. Contributes to the overall pain management strategy.
      
      ![Venn Diagram]
      
      Pre-emptive analgesia
      Regional/Local Blocks
      Multi-modal Analgesia
      
   B. Decreases inhalant requirement.
      1. Decreased cardiovascular depression associated with general anesthetics.
      2. Inhalants are expensive, so the less inhalant used means more money saved.
   C. Minimal systemic side-effects
   D. Lower post-operative analgesic dose requirements.
   E. Inexpensive
      1. Most drugs used for these particular procedures are very inexpensive.
      2. The equipment and supplies necessary for local and regional blocks are minimal and generally inexpensive.
   F. Brings in additional revenue.
      1. Charge for each additional procedure/block
      2. Clients love that you are doing everything possible to make their pet more comfortable.

II. Basic physiology of pain.
   A. Nociception – consists of three distinct physiologic processes, and perception, that are subject to pharmacologic modulation. The end result of nociception is the conscious perception of pain.
      1. Transduction – translation of physical energy (noxious stimulus) into electrical activity at the peripheral nociceptor. Nociceptors are sensitive to heat, cold, pressure, motion, touch, weight, stretch and chemical substances.
      2. Transmission – nerve impulses travel through the nervous system towards the brain.
         1. myelinated A-delta fibers conduct fast pain
         2. non-myelinated C fibers conduct slow, dull pain
      3. Modulation – endogenous analgesic systems help inhibit the stimulation of the pain pathway within the dorsal horn of the spinal cord, which changes/alters perception.
4. Perception – results from successful transduction, transmission and modulation of pain, producing the final conscious, subjective and emotional experience of pain.

III. Analgesic drugs used in local and regional blocks.
   A. Opioids – act primarily at pre- and postsynaptic receptors present in the peripheral and central nervous systems.
      1. The opioid receptors are typically classified as mu (μ), delta (δ), and kappa (κ).
      2. Activation/modulation of opioid receptors inhibits the release of excitatory neurotransmitters (glutamate, substance-P) in the dorsal horn of the spinal cord.
      3. Modulation of opioid receptors results in supraspinal, spinal and peripheral analgesia.
      4. Side effects and contraindications
         a. Use with caution in patients with cranial hypertension. Monitor respiration and be prepared to mechanically ventilate if necessary.
         b. Typically increase vagal tone resulting in reduced heart rate and in cats may increase body temperature.
         c. May cause vomiting and is contraindicated if an increase in intraocular, intracranial, intra-abdominal or esophageal pressures will be detrimental.
         d. Some animals may become dysphoric following opioid administration.
      5. Examples of some opioids are morphine, hydromorphone, oxymorphone, butorphanol, buprenorphine and fentanyl.
   B. Local anesthetics – block the initiation and conduction of electrical activity in nerves.
      1. Block sodium ion channels in neuronal cells and other tissues, thereby preventing an influx of sodium ions, membrane depolarization and creation of a propagated action potential.
      2. Analgesia is a direct result of sodium ion channel blockade and membrane stabilization.
      3. They are most frequently used to produce analgesia by administering them at specific sites (topical, local) or on nerves (regional).
      4. Local anesthetics can reduce requirements for general anesthetics. Combinations of local anesthetics with opioids have been used for epidural and intra-articular anesthesia resulting in enhanced and prolonged analgesia.
      5. Addition of epinephrine (1:200,000) delays absorption and prolongs local anesthetic action.
      6. Side effects and contraindications
         a. Large doses of local anesthetics may lead to toxic symptoms.
         b. Bupivicaine can cause cardiovascular toxicity following IV administration.
         c. Lidocaine may be safely administered IV, however, at high doses it can cause central nervous system and cardiovascular toxicity.
d. Central nervous system toxic effects include sedation, nausea, ataxia, nystagmus and tremors. Cardiovascular effects typically follow nervous system toxicity.

e. Toxic doses of local anesthetics will vary depending on route of administration, co-administered drugs such as epinephrine, site of injection and species.

f. **Toxic doses:**
   - Lidocaine – 10mg/kg in the dog, and 6mg/kg in the cat.
   - Bupivicaine – 3mg/kg in the dog and 2mg/kg in the cat.

IV. Local/regional anesthesia – the terms imply that a region of the body is affected as opposed to the entire body, as with general anesthesia.

**EPI DURAL**

**Indications for use** - Hindlimb orthopedic and soft tissue procedures (i.e. TPLO, hindlimb fracture repairs, anal saculectomy, perineal urethrostomy), laparotomy and thoracotomy.

**Materials**
- Sterile gloves
- 3ml syringe
- 6ml syringe
- Sterile 20 ga. Needle
- Spinal needle (appropriate gauge and length) – 20 and 22 gauge beveled spinal needles of various lengths are commonly used.
- 1 single dose vial sterile 0.9% sodium chloride

**Drugs and Dosages** *(Drugs marked with an * are the drugs most commonly used at WSU)*

- Preservative-free morphine (1mg/ml) – 0.1mg/kg*
  - results in a 6-18 hour duration of analgesia when given alone epidurally, and lasts much longer when co-administered with a local anesthetic (12-24 hours).
- Bupivacaine (5mg/ml) – 0.5mg/kg*
  - provides 4-6 hours of analgesia
- Lidocaine (20mg/ml) – recommended dose 1.5mg/kg
  - provides 1-3 hours of analgesia
- Preservative-free fentanyl – 5-10mcg/kg in dogs
- Preservative-free buprenorphine – 5-20mcg/kg in dogs, 5-10mcg/kg in cats.

When figuring a dose for an epidural, work out the volume of morphine, then make up the rest of the maximum volume for your patient with local anesthetic, ensuring that the local anesthetic given does not exceed the toxic dose.

**Total volume administered epidurally in dogs should not exceed 1ml/5kg, or a maximum volume of 6mls. The maximum volume in cats should not exceed 2mls.**

  - in animals where the total volume would exceed 6mls, it is recommended that the volumes of drug are reduced so as not to exceed 6mls.
**Example:** 65kg Great Dane- original dose would be 6.5mg PF morphine (6.5mls) and 32.5mg bupivicaine (6.5mls). Reduced dose would be 4mg PF morphine (4mls) and 10mg of bupivacaine (2mls) OR 6mg PF morphine (6mls) alone.

**Technique – Hanging Drop** – sterile technique is mandatory!!! (Figure 1 and Figure 2)

1. Place sedated or anesthetized animal in sternal recumbency with hind legs pulled forward.
2. Palpate the cranial edge of the wings of the ilium with your thumb and middle finger. A line connecting these two points typically over lies the spine at L7. With your index finger palpate just caudal to L7, this should be the lumbo-sacral junction, it is typically a distinct indentation, this is your site of injection. However, in overweight animals or animals with injuries to the pelvis, it can be very difficult to indentify the L-S junction.
3. Clip and aseptically scrub an area slightly larger than the area between the cranial edge of the wings of the ilium and down to the base of the tail.
4. Utilizing sterile technique open gloves and materials. Put the gloves on, maintaining sterility. Have an assistant give you the morphine into one of your 6ml syringe, if using bupivacaine do not mix your morphine and bupivacaine until you are sure there is no blood or CSF in your stylet. Then draw up 2-2 ½ mls of saline into the 3ml syringe. Maintain sterility throughout this time.
5. Utilizing sterile technique, palpate and find the L-S junction once again.
6. Needle insertion is made directly over the depression formed by the L-S junction, with the needle lined up with the spine, positioned perpendicular to the skin, and the bevel facing cranially.
7. When placing the spinal needle there will be a total of three distinct tissue layers that are passed through. Slowly insert the needle through the skin and into the muscle layer, then stop, these are the first two of the three tissue layers.
8. Remove the stylet from the spinal needle and place in a sterile area (typically the paper glove liner). Place several drops of the sterile saline in the hub of the spinal needle, forming a meniscus.
9. Steady spinal needle with fingers, check that needle is still perpendicular to the skin, bevel is forward and the needle is still in line with the spine.
10. Slowly advance the spinal needle until a “pop” is felt and/or the saline in the hub is sucked into the needle. You have passed ligamentum flavum, the third of the three tissue layers and you are now in the epidural space.
11. Using the 3ml syringe that has been previously filled with sterile saline, add air to the syringe, creating a large bubble close to the plunger of the syringe. Attach the syringe to the hub of the spinal needle and slowly inject the saline. It should flow smoothly into the epidural space and the air bubble should NOT compress. If compression occurs, you are probably not in the epidural space.
12. Once you have checked that you are in the epidural space, attach the 6ml syringe that was previously filled with your drug of choice using sterile technique, again add air to the syringe, creating an air bubble near the plunger of the syringe. Slowly inject the drug in the same manner that the saline was injected in Step 11.
13. Once the entire volume has been injected, leave the syringe attached to the needle and pull both out of animal. Pat yourself on the back, you have just completed your epidural!

***If at anytime during the procedure you get blood or CSF in the spinal needle, do not include bupivicaine or lidocaine. It is also recommended that you cut your morphine dose in half.***
Epidurals may be performed in lateral recumbency, especially in those patient’s where it may complicate their injury by placing them in sternal recumbency. When doing an epidural in lateral recumbency the hanging drop technique may still be used, however the saline may not be sucked into the epidural space upon penetration of the ligamentum flavum. If you do not want to do the hanging drop technique in lateral recumbency, the stylet may be left in the needle until the characteristic “pop” is felt when the ligamentum flavum is punctured.

Urinary retention may occur following epidural injection of local anesthetics and opioids. Patients should be periodically checked for bladder distention and catheterized if necessary.

**Contraindications**
1. Skin disorders/infections at the site of injection.
2. Clotting disorders
3. Septicemia
4. Pelvic fractures – if the fracture distorts the conformation of the epidural space, or if there inflammation or other pathology directly above the L-S junction.

**BRACHIAL PLEXUS BLOCK**

**Indications for use** - Procedures on the front limbs at the elbow or lower. This blocks the distal foot, up to the elbow region.

**Materials**
Sterile gloves
Sterile spinal needle (appropriate gauge and length) – 20 and 22 gauge beveled spinal needles are commonly used, length should be appropriate to patient size.

**Drugs and Dosages**
Bupivacaine (5mg/ml) – 1ml/4.5kg (conservative dose) or 1.5mg/kg.  
- provides 4-6 hours of analgesia
Lidocaine (20mg/ml) – 1ml/4.5kg  
- provides 1-3 hours of analgesia
*Do not exceed the toxic dose recommendation of either local anesthetic.  
*If doing bilateral blocks, figure total dose, then split the volume in half. Administer one half of total dose to each side.

**Technique (Figure 3)**
1. Place anesthetized animal in lateral recumbency, the limb that is to be blocked should be uppermost. Example, if you are blocking the right forelimb, the animal should be in left lateral recumbency.
2. Palpate the point of the shoulder, the first rib and the transverse processes of the cervical vertebrae.
3. With the neck in a natural position the cervical transverse processes will form a line which if continued will usually cross the proximal brachial plexus. With the leg also in a natural position, palpate the point of the shoulder, and the most caudal cervical vertebrae, imagine a line being drawn from each point intersecting at the first rib, this should be the area of the
brachial plexus. At the point of the triangle, imagine a third line coming straight out that is proximal to the point of the shoulder. This is your injection site.

4. Clip and aseptically prep the area cranial and dorsal to the point of the shoulder.
5. Utilizing sterile technique open gloves and materials. Put the gloves on, maintaining sterility.
6. Utilizing sterile technique, palpate and find the area described in point two.
7. The spinal needle should be inserted underneath the scapula until the tip is just caudal to the first rib. Keep the needle guided as close to the scapula as possible, you may even feel the needle scrape the bone.
8. Remove the stylet from the spinal needle and attach the syringe filled with your local anesthetic.
9. Aspirate to ensure that you have negative pressure and no blood. If you get air back in your syringe, it is possible that you have gone into the thorax, leaving the syringe attached, pull out your needle immediately. If you get blood in your syringe, there is a chance for systemic injection of the local anesthetic, which can be toxic and fatal. Again, leaving the syringe attached pull the spinal needle out. If either of these instances occur, it is best to abort the brachial plexus block and rely on other methods of analgesia for the procedure.
10. Once ensuring that you are in neither a blood vessel nor the thorax, mentally divide your total dose into three parts. Slowly administer one third of the dose at this site.
11. Slowly pull the needle out one third of the way, again aspirate to ensure your needle placement, and inject the second third of your dose. Repeat this procedure until the needle is just ready to exit the skin.
12. Once all the local anesthetic has been administered, pull the needle out with the syringe attached.

Contraindications
1. Skin disorders/infections around the site of injection.
2. History of sensitivity to local anesthetics.
3. Guard against an accidental insertion of needle into the thoracic cavity or blood vessel.

RADIAL/ULNAR/MEDIAN NERVE BLOCK (RING BLOCK)

Indications for use – procedures of the foot, such as declaw and tendonectomy.

Materials
Sterile 1cc syringe
Sterile 25ga. needle

Drugs and Dosages
Lidocaine – 0.1-0.3mls per injection site – do not exceed toxic dose!
Bupivicaine – 0.1-0.3mls per injection site – do not exceed toxic dose!

Technique – Distal technique (Figure 4)

Prep foot for declaw, cleaning with scrub and alcohol (if using a laser for the procedure, do not use alcohol).

Dorsal View
1. Palpate the carpus on the dorsal aspect of the foot.
2. Injection site is just proximal to phalanx I. This will block the superficial branches of the radial nerves.
3. Subcutaneously inject 1/3 of total dose (0.1-0.3mls). Be sure to aspirate for blood prior to injecting local anesthetic.

Palmar View
1. The first injection site is between phalanx I and the accessory carpal pad. This blocks the superficial branch of the median nerve and palmar branch of the ulnar nerve.
2. Subcutaneously inject the second third of the total dose (0.1-0.3mls). Be sure to aspirate for blood prior to injecting local anesthetic.
3. The second injection site is just proximal to the accessory carpal pad from the medial aspect. This blocks the dorsal branch of the ulnar nerve.
4. Subcutaneously inject the last 1/3 of the total dose (0.1-0.3mls). Be sure to aspirate for blood prior to injecting local anesthetic.

Contraindications and Complications
1. Skin disorders/infections around the site of injection.
2. History of sensitivity to local anesthetics.
3. Although extremely rare, patients should be observed for signs of self-mutilation following this procedure.

DENTAL NERVE BLOCKS

Materials
Sterile 1cc syringe
Sterile 22ga. or 25ga. needle
Clean latex gloves
It is suggested that each area is clipped and aseptically prepped, however it is not required.

Drugs and Dosages
Dose selection depends on the size of the animal. If multiple sites are to be injected keep the total dose below the toxic level for that species.

Lidocaine (20mg/ml) – 0.25 - 1.0 ml per site.
Bupivicaine (0.5%) – 0.25 -1.0ml per site.

Technique – Infraorbital Block – Blocks upper lip and nose, roof of nasal cavity and related skin ventral to the infraorbital foramen. (Figure 5 and Figure 7)
1. The infraorbital foramen is palpated above the third upper premolar on either side. The length of the infraorbital canal is estimated by palpating the caudal ventral margin of the bony orbit.
2. The injection site is the foramen felt dorsally to the upper third premolar. Insert the needle in a caudal-dorsal direction, holding the syringe parallel to the long axis of the jaw.
3. Insert the needle to the caudal ventral margin of the bony orbit. It should advance with very little resistance.
4. Aspirate for blood, fluid or air, if you get negative pressure then slowly inject entire volume of drug.
Technique – Mandibular Nerve Block - Blocks cheek teeth, canines, incisors, skin and mucosa of the chin and lower lip. (Figure 6)
1. Palpate the mandibular foramen from the inside of the mouth, just caudal to the last molar. This can be hard to palpate, so it is sometimes easier to palpate the nerve under the mucosa and follow it to the mandibular foramen.
2. Place index finger inside the mouth and place it on the mandibular foramen. Introduce the needle through the skin with your other hand, aiming for your index finger. Direct the needle to the foramen by palpation and when it feels properly placed, aspirate for blood, air or fluid and inject the entire volume of drug.

Technique – Mental Nerve Block – Blocks the lower lip. (Figure 7)
1. Palpate the mental foramen at the level of the second premolar.
2. Insert the needle over the mental nerve, rostral to the mental foramen, aspirate for blood, air or fluid and inject entire volume of drug.

Complications and Contraindications
1. Infected tissue
2. Animals may be at risk for self-mutilation following sensory loss to the tongue or lips.
3. Some animals may become anxious at the loss of sensation to the mouth and require sedation or even general anesthesia to offset this effect.