PRACTICAL MANUAL
OF
VETERINARY MEDICINE
(Unit I- GENERAL)

B.V.Sc. & A.H (MSVE 2016)

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CERTIFICATE

This is to be certify that Mr./Ms.…… ……..……Enrollment No…… …… … ……of Fourth year B.V.Sc. & A.H. has successfully completed all practical's in the course entitled "Veterinary Medicine" during the academic year ……

Date :
Place : CHOMU, JAIPUR

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FOREWORD

It seems interesting and delighted while going through the Practical Manual entitled "Practical Manual of Veterinary Medicine" prepared by Dr. R. K. Tanwar, Dr. D. B. Sarode, , Dr. Dhruba Das, Dr. Leenu Tanwar and Dr. Shivangi Pandey faculty in the Department of Veterinary Medicine, MJF College of Veterinary & Animal Sciences, Chomu, Jaipur (RAJ.) The Manual covers the practical syllabus of undergraduate course Prescribed by veterinary Council of India (MSVE 2016) for B.V.Sc. & A.H. programme.

This manual is written in simple language to help students to get acquainted with an approach to the various activities like clinical and practical approach to diagnose various general condition and status of body of animals

I hope this manual will make its own place in the libraries of Agricultural Universities, Veterinary and Animal Science College in near future.

I congratulate the authors for the efforts put in bringing out his practical manual.

Dean

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ACKNOWLEDGEMENT

Ever since the introduction of new course for professional B. V. Sc. & A.H. degree under Veterinary Council of India pattern in Veterinary Colleges/Universities in the country, there is a need to have a practical manual on Veterinary Medicine subject which covers the practical syllabus of undergraduate. These new course is nowdeveloped in most of the Veterinary College/University after the introduction of Veterinary Council of India programme. The present manual covers the practicaland clinical approach towards investigation and differential diagnoses of various general and systemic diseases in domestic, companion, zoo and wild animals and also includes objectives, materials required, procedures and steps to be followed towards diagnosing of diseases and organs. The main objective of this manual is to fulfill need of students and teachers teaching this course. We hope that users will find the manual immensely useful.

We look forward to receiving the valuable suggestions of readers for improvement of this manual.

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Unit 1: General

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COLLECTION OF HISTORY AND GENERAL CLINICAL EXAMINATION

Clinical examination is a fundamental part of the process of veterinary diagnosis. It provides the information required to determine the disease or diseases producing the clinical abnormalities. In addition, the information derived from the clinical examination should assist in determining the severity of the pathophysiological processes present. Without a proficient clinical examination and an accurate diagnosis, it is unlikely that the treatment, control, prognosis and welfare of animals will be optimized. The organs or systems involved, the location, type of lesion present, the pathophysiological processes occurring and the severity of the disease can be deduced from the information gained during the clinical examination. The success of clinical examination relies heavily on the knowledge of the clinician. Many clinicians begin their examination by performing a general examination which includes a broad search for abnormalities. The system or region involved is identified and is then examined in greater detail using either a complete or a problem oriented examination. For this sound knowledge of Anatomy, Physiology, Pathology and Animal behavior, skills in the methods and techniques of clinical examination, knowledge of etiology, clinical signs and pathogenesis of the diseases are the basic requirements for clinician to make diagnosis.

Taking of patient history

History taking or anamnesis is the process of obtaining information on the animal patient about its illness, onset of illness and feeding practice through careful questioning of the owner. In Veterinary practice, the disease is presented indirectly in the form of a complaint by the owner or the attendant. Thus, it is very necessary to have all the information from the owner. Most of the time, the owner or attendant fails to provide pertinent and adequate history and inaccurate history may lead to misdiagnosis. The clinician must substantiate these with rational questions utilizing professional knowledge. The disease problem of animals is difficult to diagnose without knowing the history of animals, the history should be taken from the owners of the patient and recording the owner‘s complaint. Disease information should include the group(s) affected, the numbers of animal affected (morbidity) and the identities of the animals affected; the number of animals that have died (mortality) should be established. In order to get the accurate and

complete history the following things should be focused; patient data, present history, past history and environmental history.

1. **Patient data**

Patient data is essential to identify the patient and it includes: Owner‘s name, Owner‘s address: postal address, telephone, peasant association, Species, breed, sex, age, name, ID No., body weight, Description including color, marking, polledness, and other identification marks of patient.

2. **Present history**

It comprises of recording the sequential events from the start of the patient chronic. Questions about physiological functions such as appetite, urination, defecation, rumination, respiration, sweating, milk production, gait, posture and also of the first symptoms shown by the animal should be asked. All these information deal with the current problem of the animal and the events associated with it. The point which going to be asked is that as follows:

a. **Locations of the problems:** Following up and attention at the complaint that a farmer has to say and from there you can tentatively say the likely system involved in that condition, for instance:

● Digestive system involvement will be shown as absence of rumination, appetite, bloat or diarrhea.

● Respiratory system involvement will be indicated presence of nasal discharge, coughing, dyspnea.

● Urinary system involvement will be manifested as frequent urination, passing red coloured or cloudy urine.

a. **Nature of illness:** The clinician should be able to assess and find out the time of onset of disease, any change in management practices and signs noticed by the farmers.

● To assess to know the duration of disease whether it is peracute, acute, subacute or chronic.

● To know number of animal diseased and morbidity rate and mortality rate of animals.

● Determine whether any drug has been given to animals, before it comes to clinic for assurance.

● And the following questions should be pointed:

● When did the farmer notice the disorder? (time) ?

➢ Did it occur suddenly/slowly? (acute /sub-acute / chronic nature)?

➢ What were the signs noticed? (anorexia/drop in milk yield/ others)?

➢ Are the animal fed / grazed in pasture / forest grazed? (getting information on management practices e.g. ketosis seen in stall fed animals, while babesiosis seen in forest grazed animals).

➢ Is there any other animal affected with similar condition in the same herd / in other farmer‘s herd in the village (to find out if the disease is rapidly spreading)?

➢ Ask if there has been any introduction of new animal to the herd / village (sick animal may have been bought from affected area and disease has started).

➢ Is the affected animal vaccinated against food-and mouth disease (FMD), anthrax, hemorrhagic septicemia (HS), Black quarter (BQ) (to find out if the animal is protected against common diseases).

3. Past history

Inquiring into the past history may help in arriving at a diagnosis. History of drenching a day or two earlier may cause aspiration pneumonia. History of past disease may be correlated to the present illness. Past history will also give idea if such condition prevailed previously in the area.

● Ask if such condition was reported previously too (reveal endemic nature of disease, or occurrence of a new disease).

● Does this occur at certain period of time? (find out the seasonal occurrence of the disease).

● Was the disease reported form other places in the locality? (area of spread / occurrence can be found out).

● Has any animal recovered from such a sickness? (to aid in prognosis).

● Is the disease restricted to certain age group / sex? (BQ is seen in animals between 1 – 3 years of age in both sexes).

Physical examination

The main aims of physical examination is to apply general inspection, palpation, percussion and auscultation methods used to detect clinical signs of abnormalities.

General inspection: It is done some distance away from the animal; sometimes go round the animal or herd/flock, in order to get the general impression about the case. Attention should be paid to the following items: (Behavior, Appetite, Defecation, Urination, Posture, Gait, Body condition, Body conformation)**,** Lesions on outer surface of the body can be observed: (Skin and coat, Nose, Mouth, Eyes, Legs and hoofs, Anus).

Palpation: Palpation is used to detect the presence of pain in a tissue by noting increased sensitivity by using fingers, palm, back of the hand, and fist, in order to get the information about the variation in size, shape, consistency and temperature of body parts and lesions, e.g., the superficial lymph nodes. The terms, which can be used to describe the consistency of parts during palpation, are:

● **Resilient**, when a structure quickly resumes its normal shape after the application of pressure has ceased (e.g., Normal rumen).

● **Doughy**, when pressure causes pitting as in edema.

● **Firm**, when the resistance to pressure is similar to that of the normal liver (e.g., neoplasia/tumor).

● **Hard,** when the structure possesses bone-like consistency (e.g., Actinomycotic lesion).

Fluctuating, when a wave-like movement is produced in a structure by the application of alternate pressure (e.g., hernia, hemorrhage/hematoma).

Emphysematous, when the structure is swollen and yields on pressure with the production of a crepitating or crackling sound.

Percussion: Method of examination in which part of body to be examined is struck with sharp blow using fingertips to produce audible sound. Sound thus emitted will indicate the nature of the tissue / organ involved for example rumen when bloated will emit drum like sound. Some of the organs that can be examined by percussion are: gastro-intestinal tract, abdomen and thorax, frontal and nasal sinuses. The objectives of percussion are to obtain information about the condition of the surrounding tissues and, more particularly, the deeper lying parts. Percussion can examine the area of the subcutaneous emphysema, lungs, rumen and rump. Sounds produced from various structures can be described as following list:

● Dull / flat – sound without resonance or echo, this type of sound can be heard on percussion of thick muscles or bone.

● Full sound – sound heard is with resonance but not booming like drum. This type of sound is heard from tissues like lungs that contain air inside.

● Tympanic sound – drum like sound can be heard and this type of sound is heard from bloated rumen, abomasum and intestine.

Immediate percussion: Using fingers or hammer directly strike the parts being examined.

Mediate percussion: Finger-finger percussion; Pleximeter hammer percussion. The quality of the sounds produced by percussion is classified as:

➢ Resonant**:** This is characteristic of the sound emitted by air containing organs, such as the lungs.

➢ Tympanic**:** The sound produced by striking a hollow organ containing gas under pressure, e.g., tympanitic rumen or caecum.

➢ Dull**:** Sound emitted by a solid organ like the liver or heart.

Modified percussion

Ballottement percussion: Tactile percussion or ballottement is method in which palpation and percussion are combined together to feel structures that cannot be felt by either of these methods applied singly. This is normally used for pregnancy diagnosis in cows when the foetus cannot be palpated through per rectum. Here a firm-pushing stroke is applied on to the uterus and the hand after pushing is kept in contact with uterus so that the foetus will bound and strike on it. While firm pushing is done, this sets fluid in uterus into motion and foetus is made to bounce. This modified percussion is used to detect late pregnancy in small ruminants, dogs and cats. And also, used for detection rebound of floated material shows pregnancy.

Fluid percussion: Used to detect fluid in the abdomen

Procedure**:** Apply a push on one side of the abdomen, percussion on the other side. The presence of wave-like fluid movement shows accumulation of fluid in the abdomen, e.g., ascites.

Auscultation

Word auscultation comes from _auscutona' meaning _to listen'. This is a technique of listening to the sounds produced from organs in the abdominal and thoracic cavities. In olden days listening to these sounds were done with naked ears. This had certain limitations like the skin on animal being dirty and infested by parasites it was not healthy for the clinicians and was difficult to keep ears in contact on animal body due to constant movement. Therefore, an instrument was later developed for this purpose and this is called stethoscope. The stethoscope was invented in France in 1816 by Rene Laennec at the Necker-Enfants Malades Hospital. The main objective of auscultation is listening to the sounds produced by the functional activity of an organ located within a part of the body. This method used to examine the lung, trachea, heart and certain parts of the alimentary tract.

Succussion (shaking)

It is the method used to determine the presence of fluid in the body cavities like thoracic and abdominal cavity. Here the animal is shaken from side to side to set fluid into motion so that audible fluid sound is produced. This is difficult in large animals and can be applied only in small animals like dogs and cats.

Clinical Examination of the Patient

General physical examination

Physical examination can be carried out by taking vital signs such as; Temperature taking, Pulse taking, Respiration taking, Capillary Refill Time (CRT), Physical body condition, Normal demeanour, Abnormal demeanor.

Temperature taking: Temperature is the measure of how hot or cold the animal body is. On the basis of the ability to regulate body temperature animals are divided into two groups via homeotherms and poikilotherms. Homeotherms are those animals including man that can regulate their temperature in relation to the environmental temperature. Poikilotherms are those animals that are unable to regulate their body temperature in relation to the environmental temperature (eg. Amphibians, Reptiles and Fishes). Heat is generated in the body via the intracellular oxidation of food and the muscular activities. It is lost via the physical process of conduction, convection, and radiation and through the evaporation, respiration and excretion.

Species	**Normal temp. in ^{O}C**	**Normal temp. in ^{O}F**
Cattle	37.5-39.5	99.5-101.5
Buffalo	37.5-39.0	99.5-102.2
Horse	37.5-38.5	99.5-100.4
Sheep/Goat	38.5-40.5	101.3-104.0
Pig	38.0-40.0	100.4-104.0
Dog	37.5-39.0	99.5-102.2
Cat	38-39.5	100.4-103.1
Camel	35.5-38.6	95.0-101.5
Chicken	40.5-43.0	104.9-109.4

Pulse taking: Pulse is defined as the regular expansion and contraction of the arterial wall caused by the flow of blood through it at every heartbeat. Pulse gives information with regard to the cardio-vascular abnormalities. It is influenced by exercise, excitement, annoyance, relative humidity, environmental temperature. Pulse can be adapted from the number of heart beats per

minute by using stethoscope in less manageable animals. The rhythm of pulse should also be noticed while taking pulse. The pulse rate can rise rapidly in nervous animals or those which have undergone strenuous exercise. In such cases the pulse should be checked again after a period of rest lasting 5 to 10 minutes.

Species	**Site**
Cattle and Buffalo	Middle coccygeal artery
Sheep, Goat, Dog and Cat	Femoral artery
Equine	External maxillary artery

Animals	**Rate/Min**
Cattle	60-90
Horse	28-42
Sheep/Goat	68-90
Pig	60-90
Dog	90-130
Cat	110-130
Chicken	200-400

Respiration taking: Respiratory movements can be observed at the right flank. Any change in the rate indicates respiratory involvement. Thoracic respiration is seen in animals suffering from acute peritonitis and abdominal respiration in pleurisy. Double expiratory movements are seen in emphysema in horses.

Types of respiration:

1. Costal respiration: In this type of respiration thoracic muscles are mainly involved and the movement of the rib cage is more prominent. It is seen in dogs and cats.

2. Abdominal respiration: This type of respiration is seen in ruminants viz cattle, goat, sheep and yak. Here the abdominal muscles are involved and movement of the abdominal wall is noticed.

3. Costo- abdominal respiration: This type of respiration is seen in horses and in this type of respiration muscles of both thorax and abdomen are involved so the movement of the ribs and the abdominal wall are noticed.

The respiration rate is measured through counting of either contraction or expansion of the thorax and abdomen which can be observed during clinical examination.

Animals	**Per Minute**
Horse	9-10
Cattle	27
Sheep/Goat	12-15
Pig	10-20
Cat	20-30
Dog	20-22
Chicken	15-30
Camel	5-12

Terms used in respiration

Eupnoea: Normal breathing

Polypnoea: Increase in respiration rate

Dyspnoea: Difficulty in respiration

Panting: Increased rate with reduced depth

Hyperpnoea: Increased rate with increase in depth

Oligopnea: Abnormally slow and shallow breathing

Apnoea: Cessation of respiration

Visible mucous membrane: The mucous membrane in the eyes, mouth and vagina (in the case of females) can be examined to determine the health status of an animal. Examination of the

- Mucous membrane examination should be done in natural light (sunlight) not in the lamplight. The abnormalities of color of mucous membrane are caused by different factors like
- Pallor of the mucous membranes may indicate anaemia caused by direct blood loss or by haemolysis,
- A blue tinge may indicate cyanosis caused by insufficient oxygen in

  the blood, A yellow colour is a sign of jaundice,
- The mucosae may be bright red (sometimes described as being _injected mucous membranes‘) in febrile animals with septicaemia or viraemia,
- Bright red colouration of the conjunctiva is often seen, for example, in cases of bovine respiratory syncitial virus infection.
- A cherry-red colouration may be a feature of carbon monoxide poisoning.
- A greyish tinge in the mucosae may be seen in some cases of toxaemia – such membranes are sometimes said to be _dirty‘.
- High levels of methaemoglobin, seen in cases of nitrate and/or nitrite poisoning, may cause the mucosae to be brown coloured.

Anaemic mucous membranes.

➢ Blood loss anaemia.

➢ Parasitic infestations leading to haemolysis.

➢ Tumours or leucosis.

➢ Iron deficiency anemia.

➢ Long-standing infectious diseases.

➢ Exposure to X-rays and some medications.

Congested mucous membranes.

- High environmental temperatures and exercise.
- Any disease resulting in fever.
- Diseases of the heart, brain and its membranes.

Yellowish or icteric mucous membranes.

➢ Icterus of jaundice occurs due to increase of blood bilirubin concentration (blood parasites, leptospirosis, hepatitis, cholangitis, cholecystitis and cholangiohepatitis).

➢ Infectious anaemia and contagious pleuropneumonia of horses.

➢ Chronic gastric dilatation.

Cyanosed mucous membranes.

➢ Bluish discoloration of visible mucous membranes resulting from presence of reduced haemoglobin in blood capillaries.

➢ Myocarditis, pericarditis.

➢ Plant and mineral intoxications.

Swelling of mucous membranes: Inflammation of mucous membranes results in its swelling; in which case the mucous membranes may also be hot and tender (i.e. showing cardinal signs of inflammation). Marked swelling of conjunctival mucous membranes is characteristic of equine influenza. A slight degree of swelling is noticed in contagious pleuropneumonia of horse and cattle plague, anthrax and fowl diphtheria.

Capillary Refill Time (CRT): Capillary refill time (CRT) is defined as –time required for return of color after application of blanching pressure to a distal capillary bed. This is taken by compressing the mucosa of the mouth or vulva to expel capillary blood, leaving a pale area, and recording how long it takes for the normal pink colour to return. In healthy animals, the CRT should be less than 2 seconds. A CRT of more than 5 seconds is abnormal, and between 2 and 5

seconds it may indicate a developing problem. An increase in CRT may indicate a poor or failing circulation causing reduced peripheral perfusion of the tissues by the blood.

Physical body condition: Body condition scoring is an important management practice used by producers as a tool to help optimize production, evaluate health, and assess nutritional status. Different scores can be given for individual animal and can further classified as normal, fatty, lean/thin, emaciation.

Condition Score 1: Very thin**:** This animal‘s skeletal structure is very prominent. Notice the deep depressions next to the spine, between the pelvis and rib cage, between the hooks and pins, and around the tail head.

Condition Score 2: Thin**:** The animal‘s skeleton is still very apparent. The individual spinous processes are clearly visible, but there is a small amount of fat tissue over the spine, hooks, and pins.

Condition Score 3: Medium (Normal body condition): The animal appears smooth over the spine, ribs, and pelvis and the skeletal structure can be easily palpated. The hooks and pins are still discernible, with a moderate, rather deep depression between the pelvis and rib cage, hooks and pins, and around the tail-head.

Condition Score 4: Fat**:** There are no spinous processes detectable, and no depression in the loin area, which gives the top-line of the animal a flat, tabletop appearance. The ribs can no longer be felt, and the pelvis can only be felt with firm pressure. The hooks and pins have a rounded appearance due to areas of fat covering.

Condition Score 5: Very Fat**:** The animal appears rounded and smooth with a square-shaped appearance, because of the amount of fat filling in the loin. The skeletal structure is no longer visible, and can only be palpated with excessive pressure.

Normal demeanour: When, on being approached, an animal makes a normal response to external stimuli, such as movement and sound, the demeanour is said to be normal (bright). Normal reaction under these circumstances may consist of elevating the head and ears, turning towards and directing the attention at the source of stimuli, walking away and evincing signs of attack or flight.

Abnormal demeanour: Behavioral change/ response to external stimuli. The Abnormal demeanours in domestic animals are as follows:

● Decreased response (depression): dull (apathetic); dummy state; comma.

● Excitation or increased response: apprehension (mildly anxious); restlessness; mania; frenzy.

● Posture: It denotes the anatomical configuration when they remain in stationary situation. How does it stand? How does it sit? How does it lie?

● Gait: It indicates about the locomotory process of an animal.

● Body conformation: shape and size of the different body regions relative to other regions.

Regional or systematic clinical examination

Clinical Examinations of the head and neck region:

Before handling the head a further visual inspection and observation of the head and neck is advisable as whether the following questions are present:

● Movements of head and neck – normal or abnormal

● Carriage of head – normal or tilted,

● Can the animal see?

● Can the animal hear?

● Ocular or nasal discharge,

● Salivation – normal or excessive,

Ability to prehend, masticate and swallow food

● Mobility of the neck.

Clinical Examinations of the thorax:

Lungs, heart and other vital organs are situated in the thorax. Inspection of thorax is important to know about respiration. Rate, rhythm, depth and type of respiration can be observed by examining the thoracic region. Place the back of hand in front of nostrils or by closely examining the movement of the thoracic cavity.

Clinical Examinations of the abdomen:

Variation in size of abdomen can be noticed. Decrease in size of abdomen is termed as gaunt. Occurs due to starvation, severe diarrhea and chronic diseases with loss of appetite. Other abnormalities of abdomen are ventral oedema, congestive heart failure, gangrenous mastitis, protein deficiency.

Clinical Examinations of the external genitalia:

In males, examination may reveal inflammatory or neoplastic enlargements of preputial sheath or scrotum, presence of abscess on prepuce or sheath and any abnormal discharges. In females, swelling of vulva and vagina, prolpase, abscess and presence of pus and blood, or retained placenta can be seen.

Clinical Examinations of the mammary glands:

Various affections of udder can be determined on the basis of palpation and examination.

Exercise: 2 **Date:**

VETERINARY LABORATORY SAMPLING TECHNIQUES

The starting point for the laboratory investigation of any animal disease is the taking of samples. Samples may be taken from animals or the environment for a variety of purposes, such as disease diagnosis, disease surveillance, health certification or monitoring the response to treatment or vaccination. The samples collected should be appropriate for the intended purpose, and adequate in number and amount to provide statistically valid results. Diagnostic laboratories require the submission of appropriate samples that arrive at the laboratory in good condition. For disease diagnosis, the tissues sampled should be representative of the condition being investigated and the lesions observed. Samples should be taken with care, to avoid undue stress or injury to the animal or danger to the operator. Where appropriate samples should be collected aseptically, and care should be taken to avoid cross-contamination between samples.

The samples should be carefully packaged, labeled, and transmitted to the laboratory by the fastest practicable method, with the appropriate temperature control. There are specific requirements for the packaging and shipping of infectious substances, including diagnostic specimens, which must be followed. If material is sent to a laboratory in another country, this laboratory should be consulted in advance to ensure that it is willing to receive the material and to obtain the appropriate import license. All samples should be accompanied by a letter or submission form, which includes the name and address of the submitter, the origin of the material, the relevant history, animal identification and corresponding specimens, and the tests requested.

A. COLLECTION OF SAMPLES

Before taking samples, careful consideration should be given to the purpose for which they are required. This will determine the type and number of samples needed to provide valid results. When samples are taken from live animals, care should be taken to avoid injury or distress to the animal or danger to the operator and attendants. It may be necessary to use mechanical restraint,

tranquillization or anaesthesia. Whenever handling biological material, from either live or dead animals, the risk of zoonotic disease should be kept in mind and precautions taken to avoid human infection. Post-mortem examinations should be carried out under aseptic conditions as is practicable. Care should be taken to avoid environmental contamination, or risk of spread of disease through insects or fomites. Arrangements should be made for appropriate safe disposal of animals and tissues.

Considerable skill and care are required to decide on the correct samples to be sent to the laboratory. The samples collected should be representative of the condition being investigated and the lesions observed. Frequently, a combination of blood samples for serology and tissues from dead or culled animals for microbiological culture will be required.

1. Sample collection from live animals

a) Blood

Blood samples may be taken for haematology or for culture and/or direct examination for bacteria, viruses, or protozoa, in which case it is usual to use anticoagulants, such as ethylene diamine tetra-acetic acid (EDTA) or heparin. They may also be taken for serology, which requires a clotted sample. Blood plasma is also used for some procedures. A blood sample is taken, as cleanly as possible, by venipuncture. In most large mammals, the jugular vein or a caudal vein is selected, but brachial veins and mammary veins are also used. In birds, a wing vein (brachial vein) is usually selected. In small laboratory animals, the vena auricularis or vena retroorbitalis may be useful to obtain blood samples or it may be obtained by heart puncture. Blood may be taken by syringe and needle or by needle and vacuum tube (not easy in delicate veins but convenient in strong veins). Small quantities of blood are conveniently obtained by pricking with a triangular, solid-pointed needle. Ideally the skin at the site of venipuncture should first be shaved (plucked) and swabbed with 70% alcohol and allowed to dry.

For samples that are collected with anticoagulant, thorough mixing, using gentle agitation only, is necessary as soon as the sample has been taken. It may also be necessary to make a smear of fresh blood on a microscopic slide; both thick and thin smears may be prepared.

For serum samples, the blood should be left to stand at ambient temperature (but protected from excessive heat or cold) for 1–2 hours until the clot begins to contract. The clot can then be ringed round with a sterile rod and the bottles placed in a refrigerator at 4°C. After several hours, or overnight, the sample can be centrifuged at about 1000 **g** for 10–15 minutes and the serum can be decanted or removed with a pipette. In order to establish the significance of antibody titres, paired serum samples will often need to be collected 14 days apart. An alternative method for collecting and transporting blood that is to be used for serology is to place a drop of blood on to filter paper, the blood is dried at room temperature and the sample can then be shipped unrefrigerated.

b) Faeces

At least 10 g of freshly voided faeces should be selected. Faeces for parasitology should fill the container and be sent refrigerated to prevent hatching of parasite eggs and should arrive at the laboratory within 24 hours. Screw top containers or sterile plastic bags should be used for shipment; avoid tubes with rubber stoppers as gas generated can result in blowing the stopper off the tube, ruining the integrity of the sample and contaminating other samples in the package. An alternative and sometimes preferable method is to take swabs from the rectum (or cloaca), taking care to swab the mucosal surface. The swabs should be visibly coated with faecal material; however, samples collected with a swab are inadequate for parasitology. Care should be taken when collecting swabs from small, delicate animals or birds to avoid injury to the animal; small swabs are commercially available that should be used. Swabs should be transported in appropriate transport medium. Faeces are best stored and transported at 4°C.

c) Skin

In diseases producing vesicular lesions, collect, if possible, 2 g of affected epithelial tissue aseptically as possible and place it in 5 ml phosphate buffered glycerin or Tris-buffered tryptose broth virus transport medium at pH 7.6. Additionally, the vesicular fluid should be sampled where unruptured vesicles are present; if possible, vesicular fluid should be aspirated with a syringe and placed in a separate sterile tube. Plucked hair or wool samples are useful for surface feeding mites, lice and fungal infections. Deep skin scrapings, using

the edge of a scalpel blade, are useful for burrowing mites and, in birds, feather tips can be taken for detection of viral antigen where Marek‘s disease is suspected.

d) Genital tract and semen

Samples may be taken by vaginal or preputial washing, or by the use of suitable swabs. The cervix or urethra may be sampled by swabbing. Samples of semen are best obtained using an artificial vagina or by extrusion of the penis and artificial stimulation. The sperm-rich fraction should be present in the sample and contamination by antiseptic washing solutions should be avoided.

e) Eye

A sample from the conjunctiva can be taken by holding the palpebra apart and gently swabbing the surface. The swab is then put into transport medium. Scrapings may also be taken on to a microscopic slide. The handles of metal-handled swabs are useful for this, to ensure that sufficient cells are removed for microscopic examination.

f) Nasal or ocular discharge (saliva, tears)

Samples may be taken with dacron, cotton or gauze swabs, preferably on wire handles as wood is inflexible and may snap. It may be helpful if the swab is first moistened with transport medium. The swab should be allowed to remain in contact with the secretions for up to 1 minute, then placed in transport medium and sent to the laboratory without delay at 4°C. Long protected nasopharyngeal swabs should be used to collect samples for some suspected viral infections.

g) Milk

Milk samples should be taken after cleansing and drying the tip of the teat, the use of antiseptics should be avoided. The initial stream of milk should be discarded and a tube filled with the next stream(s), a sample of bulk tank milk can be used for some tests. Milk for serological tests should not have been frozen, heated or subjected to violent shaking.

2. Sample collection at post-mortem

Samples of tissue from a variety of organs can be taken at post-mortem. Animal health personnel should be trained in the correct procedures for post-mortem examination of the species of animals with which they work. The equipment required will depend on the size and species of animal, but a knife, saw and cleaver will be required, and also scalpel, forceps and scissors, including scissors with a rounded tip on one blade, for opening intestines. A plentiful supply of containers appropriate to the nature of the sample required should be available, along with labels and report forms. Containers should be fully labeled with the date, tissue and animal identification. The operator should wear protective clothing: overalls, washable apron, rubber gloves and rubber boots. Additionally, if potential zoonotic diseases are being investigated, the post-mortem examination should be conducted in a biological safety cabinet; if this is not possible, an efficient face mask and eye protection should be worn. If rabies or transmissible spongiform encephalopathies (TSEs) are suspected, it is usual to detach the animal‘s head.

Tissues may be collected for microbiological culture, parasitology, biochemistry, histopathology and/or immuno-histochemistry, and for detection of proteins or genome nucleic acids. The person conducting the post-mortem examination should have sufficient knowledge of anatomy and pathology to select the most promising organs and lesions for sampling. Each piece of tissue should be placed in a fully labeled separate plastic bag or sterile screw-capped jar. Sterile instruments should be used for collecting specimens for microbiological culture and care should be taken not to contaminate tissues with intestinal contents. Disinfectants should not be used on or near tissues to be sampled for bacterial culture or virus isolation.

The tissues may be sent to the laboratory dry or in bacterial or virus transport medium, depending on the examinations required. After collection, the samples for microbiological examination should be refrigerated until shipped. If shipment cannot be made within 48 hours, the samples should be frozen; however, prolonged storage at –20°C may be detrimental to virus isolation. For histopathology, blocks of tissue not more than 0.5 cm thick and 1–2 cm long are cut and placed in neutral buffered 4–10% formalin, which should be at least ten times the volume of the tissue sample. For certain suspected diseases, larger portions of brain are required; the brain is sectioned using a sagittal cut, half is submitted fresh, on ice, and the other half is

submitted in 10% buffered formalin. Store and pack formalin-fixed tissues separately from fresh tissues, blood and smears. Care should be taken to insure that formalin-fixed tissues are not frozen. Once fixed, tissues can be removed from formalin and, as long as they are kept moist and protected (e.g. by wrapping in formalin-soaked paper towels, then sealed in screw-capped jars), they can be forwarded to the laboratory without formalin.

3. Environmental and feed sampling

Samples may be taken to monitor hygiene or as part of a disease enquiry. Environmental samples are commonly taken from litter or bedding and voided faeces or urine. Swabs may be taken from the surface of ventilation ducts, feed troughs and drains. This kind of sampling is particularly important in hatcheries, artificial insemination centers and slaughter houses in which specialized equipment is maintained. Samples may also be taken from animal feed, in troughs or bulk containers. Water may be sampled in troughs, drinkers, header tanks or from the natural or artificial supply.

B. INFORMATION TO BE SENT ALONG WITH THE SAMPLES

It is essential that individual samples be clearly identified using appropriate methods. Marking instruments should be able to withstand the condition of use, i.e. being wet or frozen. Pencil has a tendency to rub off containers and labels attached to plastic will fall off when stored at –70°C. Information and case history should always accompany the samples to the laboratory, and should be placed in a plastic envelope on the outside of the shipping container. The following are suggested items that should be addressed. It would be advisable to contact the receiving laboratory to determine if it has a submission form that it would like to have submitted with the samples or if it needs other information.

i) Name and address of owner/occupier where disease occurred, with telephone and fax numbers.

ii) Name, postal and e-mail address, telephone and fax numbers of the sender.

iii) Diseases suspected and tests requested.

iv) The species, breed, sex, age and identity of the animals sampled.

v) Date samples were collected and submitted.

vi) List of samples submitted with transport media used.

vii) A complete history would be beneficial for the laboratory and should be included if possible. Some of the components of the history are:

a) A list and description of the animals examined and the findings of the post-mortem examination.

b) The length of time sick animals has been on the farm; if they are recent arrivals, from where did they originate.

c) The date of the first cases and of subsequent cases or losses, with any appropriate previous submission reference numbers.

d) A description of the spread of infection in the herd or flock.

e) The number of animals on the farm, the number of animals dead, the number showing clinical signs, and their age, sex and breed.

f) The clinical signs and their duration including the condition of mouth, eyes and feet, and milk or egg production data.

g) The type and standard of husbandry, including the type of feed available, possible contact with poison or poisonous plants.

h) Any medication given to the animals, and when given.

i) Any vaccination given, and when given.

j) Other observations about the disease and husbandry

C. PACKAGING AND TRANSPORT OF SAMPLES

1. Approval to ship specimens

The laboratory that is going to receive the samples should be contacted to ensure that it has the capability to do the testing requested and to see if there are any special packaging or shipping requirements. It is essential to contact the receiving laboratory when material is sent to another

country. A special import license will usually be required and must be obtained in advance for any biological material. This license should be placed in an envelope on the outside of the parcel.

2. Transportation of specimens

The specimens should be forwarded to the laboratory by the fastest method available. If they can reach the laboratory within 48 hours, samples should be sent refrigerated. If dry ice is used, the additional packaging requirements must be met. Infectious substances, which can include diagnostic specimens, are not permitted to be shipped as checked luggage or as carry on luggage and must be shipped as cargo.

3. Packaging

The shipper should ensure that the specimens are packaged so they arrive at the laboratory in good condition and there is no leakage during shipment. The International Air Transport Association (IATA), Dangerous Goods Regulations (DGR) has explicit requirements for packaging and shipment of diagnostic specimens, by all commercial means of air transport. In many countries, similar requirements are applicable to ground shipments and the postal service.

The IATA lists the following exemption from the Dangerous Goods Regulations:

Substances which do not contain infectious substances or substances which are unlikely to cause disease in humans or animals are not subject to these regulations unless they meet the criteria for inclusion in another class.

There are also exceptions for some Biological Products and the shipper of these products is referred to the IATA Regulations for these requirements as some Biological Products are not exempted.

Exercise: 3 **Date:**

NASOGASTRIC INTUBATION

Nasogastric intubation, more commonly known as stomach tubing, involves passing a hollow tube up the horse‘s nose, down the oesophagus (gullet) into the horse‘s stomach. It is used by a vet to identify if there are any abnormal contents in the horse‘s stomach, and to administer fluids and some treatments directly into the stomach. It is the second most commonly used test to help diagnose horses with colic (rectal examination being the most common)**.** Horses may resent this action being undertaken, but it can be an essential procedure to perform in some cases.

Why is it performed?

The anatomy of the horse‘s gastrointestinal tract means that in theory, horses are unable to vomit. Therefore, any blockages resulting in a build-up of food and fluid within the stomach or small intestine can‘t be relieved. Instead the stomach becomes more and more distended (swollen), causing severe pain. If this distension is not relieved, the stomach can rupture with fatal consequences. This is rare and only occurs with certain types of colic, but it is an important reason why a vet may want to use a stomach tube. Nasogastric intubation is also the only method of giving fluids directly into the intestinal tract. Fluids are used to relieve conditions such as impactions, to provide electrolytes and administer other treatments as required. This can be an important part of treatment as horses with colic are often reluctant to eat or drink.

Limitations and possible complications

There are some potential complications of nasogastric intubation; the most common resulting in the horse having a nose bleed. There is a highly vascular (lots of blood vessels) structure called the ethmoturbinates at the back of the horse‘s nasal cavity which the stomach tube passes next to. As a result it is not uncommon for this to bleed during or after intubation. Nose bleeds in a horse can look quite dramatic; it can look like a large amount of blood, even though it is a small proportion of their total blood volume (for example, an average 500kg horse has over 50 litres of blood in its body). The sensation of a nosebleed will also make the horse blow out through their nose, spreading the blood over their surrounding area. Although nose bleeds in the horse can look dramatic, they are not painful to the horse, and should stop without any problems given

enough time. In some horses it can be difficult to pass the tube into the oesophagus and repeated attempts may be required. Horses that are sedated or are very sick may have a reduced swallow response making it harder to pass the tube. It is very important that the tube is passed into the oesophagus and not the airway and therefore your vet will take extra time to check the position of the tube.

GASTRIC INTUBATION, GAVAGE, LAVAGE

Synonyms

Gastric decompression, orogastric feeding, orogastric intubation

Overview and Goals

Passage of a hollow tube into the mouth and through the oropharynx into the stomach to facilitate decompression of gas, removal of stomach contents (lavage), or administration of large volumes of liquid, food, or medication (gavage)

Indications

• Gastric intubation:

○ Preoperative stabilization of gastric dilatation/volvulus (GDV); allows evacuation of gas and fluid, resulting in an improved hemodynamic state

○ Relief of discomfort associated with gaseous dilatation (without torsion) of the stomach

• Gavage:

● Administration of large volumes of liquid medication, including:

▪ Activated charcoal after toxin ingestion

▪ Barium for gastrointestinal (GI) contrast radiography

▪ Hyperosmotic laxative agent prior to colonoscopy

● Administration of formula to neonatal animals that are not nursing on their own

• Lavage:

● Preoperative stabilization of GDV. Removal of stomach contents may help decrease the speed of gas reaccumulation while the animal is being prepared for surgery, thus slowing or preventing cardiovascular deterioration.

● Removal of stomach contents with suspected intoxications

Note: Gastric lavage may not be indicated in all cases of toxin ingestion. Substance ingested, consistency, time since ingestion, and animal status will influence whether gastric lavage is appropriate.

Contraindications

• Esophageal disease that could lead to tube-induced trauma or perforation. Conditions of concern include esophageal stricture, neoplasia, ulceration, megaesophagus, and recent esophageal surgery.

• Gastric disease that could lead to tube-induced trauma or perforation.

• Any swallowing disorder (megaesophagus, esophageal motility disorder, etc.), pharyngeal disorder, or laryngeal disorder (paralysis, previous tie-back surgery, etc.) that could predispose a nonendotracheally intubated animal to aspiration.

• If even one of these conditions is present, the risk of the procedure versus its benefits must be considered (and will vary from case to case) before deciding whether to perform the procedure.

Equipment, Anesthesia

Gastric intubation:

• Two assistants (minimum)

• Flexible plastic tubing of various length and diameter. The distal end must be smooth and atraumatic; smoothing may be achieved by brief heating of the end of the tube over a flame, cooling, and trimming edges with a scalpel blade. One to three side holes may facilitate

evacuation of stomach contents by minimizing obstruction of a single distal hole with gastric mucosa or ingesta.

• A roll of clinic-type white cloth tape

• Water-soluble lubrication jelly

• Mouth gag/speculum

If gastric lavage, all of the above, plus:

• Funnel or stomach pump

• Container (e.g., bucket) to collect stomach contents and lavage fluid

• Lavage fluid: usually warm (body temperature) water

Anticipated Time

Dependent on cooperation of animal; additional time may be needed for sedation or general anesthesia:

• Gastric intubation: 2-5 minutes

• Gavage: 3-10 minutes

• Lavage: 10-60 minutes

Preparation: Important Checkpoints

• Ensure that adequate manual or chemical restraint for the procedure is planned. Personal preference and animal stability may dictate the degree of sedation or anesthesia chosen. Note: Some clinicians prefer to ensure a patent and protected airway to minimize the potential for aspiration pneumonia through the use of general anesthesia and a cuffed endotracheal (ET) tube when gastric lavage is performed.

● Maximize cardiovascular stability prior to the procedure.

Possible Complications and Common Errors To Avoid

● Inadvertent passage of the orogastric tube into the trachea can result in mild to severe complications:

● Tracheal irritation leading to transient coughing or mucosal bleeding is possible.

● Tracheal or bronchial placement of the gastric tube can result in airway obstruction until the tube is repositioned.

● Tracheal or bronchial tearing can result in pneumomediastinum, pneumothorax, and death.

● Tracheal or bronchial administration of gavage or lavage fluids can result in severe aspiration pneumonia and death.

• Oral, pharyngeal, laryngeal, esophageal, or gastric trauma can result if excessive force is used for passing the gastric tube. Full-thickness tearing is possible, especially with a preexisting underlying disease.

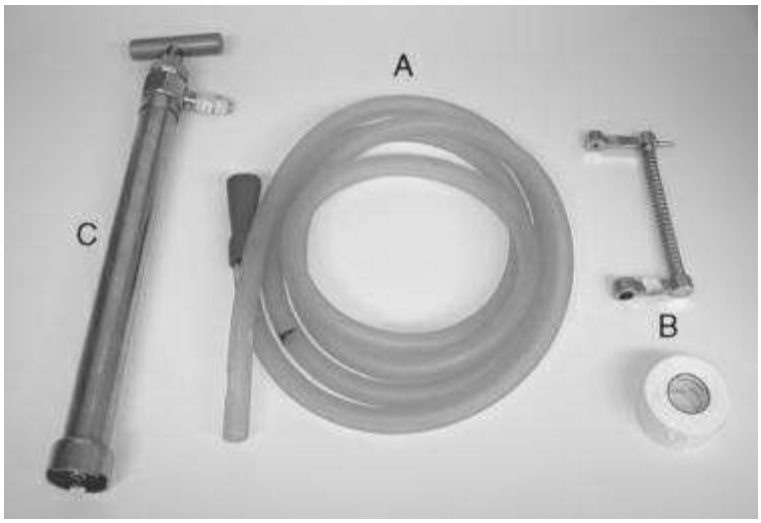
• Inability to pass the tube into the stomach may be due to the choice of a tube with a diameter that is too large, esophageal obstruction (foreign body, stricture, neoplasia), torsion of the stomach, or excessive lower esophageal sphincter (LES) tone. Discontinuation of metoclopramide prior to elective gastric intubation is recommended to minimize LES tone.

• Inadequate sedation of an uncooperative animal will lead to longer procedure times and increased risk of injury to the animal and veterinary staff.

• Inability to effectively remove gastric contents through lavage may be related to excessive size or adhesive nature of gastric contents, gastric compartmentalization, or other factors.

• Regurgitation during lavage, gastric overfilling, or esophageal administration of large volumes of lavage fluid can result in aspiration if a cuffed ET tube is not in place.

• Excessive tube advancement can cause occlusion of the distal end of the tube against stomach mucosa. Palpation of the tube pressing against the stomach wall may indicate a need for partial retraction.



Materials and equipment used for gastric intubation and lavage. **A**, Orogastric tube. **B**, Metal speculum or roll of tape to be used as a mouth gag. **C**, Stomach pump for lavage.

Procedure

• Manual restraint, sedation, or general anesthesia as indicated

• Position animal in sternal recumbency. If animal is uncomfortable, alternate positions may be better tolerated (sitting, standing, lateral, etc.).

○ Placement of the animal on an elevated surface will allow gravity-assisted efflux of stomach contents and lavage fluid once the tube is in place.

• Choose appropriate tube diameter for esophageal size and procedure planned. Example: A tube with an outer diameter of 1.5 inches (3.5 cm) is appropriate for most medium-sized dogs (45 lb [20 kg]). A larger tube size may be necessary for effective lavage versus gas decompression.

• Measure the length of tube necessary to pass from the nose to the xiphoid. Mark this distance on the tube with a piece of tape or nontoxic marker.

• Place a mouth gag (speculum) to prevent the animal from chewing on the tube.

○ A roll of 2-inch (5 cm)-wide clinic-type white cloth tape works well in many animals. The tube will pass through the hole in the tape roll. Place tape roll on top of the tongue and behind all 4 canine teeth.

○ Have an assistant hold the mouth closed around the mouth gag.

○ Avoid using a gag that will damage the teeth.

• Generously lubricate the distal portion of the stomach tube.

• Pass the tube into the mouth through the mouth gag.

Exercise: 4 **Date:**

OXYGEN THERAPY IN VETERINARY PRACTICE

Oxygen therapy denotes the delivery of high concentrations of oxygen into the respiratory system to increase the oxygen levels in the blood so that more oxygen reaches at tissues level. It is a key component in respiratory care. The body constantly takes oxygen and releases carbon dioxide and when this process is inadequate, oxygen levels in the blood falls and the patient may need supplemental oxygen. The purpose is to increase oxygen saturation in tissues where the saturation levels are too low due to illness or injury.

Indications of Oxygen Therapy

Hypoxia

Hypoxia, which constitutes the most important indication for oxygen therapy, is defined as lack of oxygen at the tissue level. Hypoxaemia, on the other hand, is low arterial oxygen tension (PaO_2) below the normal levels. There are numerous clinical conditions which could cause hypoxemia, as discussed in the following conditions-

Decreased fraction of inspired oxygen: Conditions like central nervous system weakness, neuromuscular weakness and diseases of thorax like pneumothorax/hydrothorax etc. interferes with the normal ventilation.

Alveolar hypoventilation: Pathological conditions like pneumonia and fibrosis of lungs results thickening of alveolar membrane which causes oxygen to be displaced by carbon dioxide in poorly ventilated alveoli.

Impairment of diffusion, due to pneumonia or fluid/pus in alveoli also results hypoxemia.

In ventricular septal defect, there is mixing of oxygenated and non-oxygenated blood in the heart which results hypoxemia.

Anaemic Hypoxia

This occurs when inadequate haemoglobin is available to transport oxygen. Conditions like

haemorrhages, chronic tick infestation, blood protozoan diseases, epistaxis, nutritional deficiency etc. results anaemic hypoxia.

Stagnant Hypoxia

Stagnant hypoxia is caused by low blood flow, inadequate oxygen delivery to the tissues, dehydration, haemorrhage, low cardiac output and CHF.

When to Institute Oxygen Supplementation

Clinical signs like nasal flaring, pallor of mucus membrane, cyanosis, panting, irregular chest wall movements etc., require immediate supplementation of oxygen. Quantitatively, oxygen should be provided to any patient with saturation of oxygen (SaO_2) or pulse oximetry reading (SpO_2) of <93% or with an arterial partial pressure of oxygen (PaO_2) of <80 mm Hg.

Use of Pulse Oximetry for Detection of Level of Oxygen Deficiency

Pulse oximetry is the most non-invasive and stress-free method of monitoring oxygen delivery to the tissues. It calculates the percent of oxygen saturation of haemoglobin in arterial blood using spectrophotometry. The probe passes light through the tissues at two different wavelengths: a red and infrared light absorption. Oxygenated haemoglobin absorbs more infrared light and allows more red lights to pass through. Deoxygenated haemoglobin absorbs more red lights and allows more infrared light to pass through. The difference in light absorption is calculated and the final figure is displayed as a percentage (PO_2%).

Methods of Oxygen Therapy

There are 2 methods of oxygen therapy used in small animals.

Non-invasive Method

Invasive Method

Non-Invasive Methods of Oxygen Therapy

Non-invasive ventilation (NIV) refers to the administration of ventilator support without using an invasive artificial airway (endotracheal tube or tracheostomy tube). This method includes-

a) flow-by oxygen

b) face mask

c) oxygen hood/oxygen collar and

d) oxygen incubator/oxygen cage.

Flow-by Oxygen

This method is the simplest among all non-invasive methods. Here, oxygen pipe placed adjacent to, or within 2cm of a patient‘s nostril or mouth. In this method there is no need of humidifier (overall fractional oxygen concentration being delivered to the lungs < 35). A flow rate of 2L to 3L/minute was maintained, which provides a FiO_2 of 25 to 40 per cent.

Face Mask

Face mask of variable sizes are widely available. It can be comfortably placed over the patient‘s muzzle. A tight-fitting seal is required to allow for optimal oxygen delivery. While using a face mask, attention should be made to ensure that there is a gap between the patient‘s nares and the oxygen delivery point and patient‘s nares should not be squashed to the end of the mask. In this method, flow rate of 2L/minute to 5L/minute can provide a FiO2 of up to 50 to 60 %. If a loose-fitting mask is used, a much higher flow rate will be required.

Complications

Tight-fitting masks may results rebreathing of carbon dioxide and an increase in temperature and humidity of intake air.

Oxygen Hood /Oxygen Collar

Oxygen hood /oxygen collar produces a very effective oxygen delivery method, using low flow rates. Commercial oxygen hoods are available in markets. It can be made easily by using a rigid Elizabethan collar and plastic wrap. An oxygen tube must be attached two inches from the base of the collar. The plastic wrap is placed across the front of the collar, covering two-thirds of the front and secured to the sides. Opening acts as a vent which releases excess oxygen and exhaled carbon dioxide. As oxygen is heavier than air, it remains in the lower two-thirds of the collar,

acting as a reservoir. Here flow rates of 0.5L/minute to 1L/minute should maintained, will deliver a FiO2 of 30 to 40 %. Heat and humidity within the hood/collar should be monitored. Also ophthalmic ointments are applied on a regular basis to prevent drying.

Oxygen Incubator/Oxygen Cage

This provides an excellent means of supplying oxygen to smaller patients within a stress-free environment. Medical pediatric incubators and commercially available portable oxygen cages/tents can be used in veterinary practice. A flow rate- 2 to 10 L/minute (depending on the size of the cage) should be maintained. It also has some disadvantages like the patients can rapidly become hyperthermic, there is rapid increment of humidity within a short time limiting its use for short periods and it restrict immediate access to the patient.

Invasive Methods of Oxygen Therapy

This technique is only possible when the animal is unconscious or under anaesthesia. It includes a) nasal oxygen prongs, b) nasal oxygen catheters and c) trans-tracheal oxygenation.

Nasal Oxygen Prongs

Nasal prongs are widely used in human medicine. Size of prongs varies from new borne, pediatrics to adults. A prong is placed at each nare and aligned with the opening of the nostril. Prongs can slip out easily, which may require securing with a suture or surgical staples. Here the flow rates ranges from 3L/minute to 6L/minute.

Nasal Oxygen Catheters

Application of nasal catheter for oxygen delivery is the best method among all the invasive method as it is inexpensive, technically easy to place and also well tolerated by the patient. Oxygen catheters are available in various diameters and lengths. Flow rates of 50ml/kg/minute to 150ml/kg/minute should be maintained (can provide a FiO2 of 30 to 70 per cent).

Transtracheal Oxygenation

This technique is used in patients intolerant to nasal oxygen delivery like patients suffering from upper airway obstruction. Catheters made up of silicones are placed surgically between the

fourth and fifth tracheal cartilaginous rings and the end of the catheter can then be attached to the oxygen source. Here the flow rate is 50ml/kg/minute, which provides 40 to 60 % FiO2.

Oxygen Toxicity

Oxygen toxicity occurs when oxygen administered in excessive amounts over a prolonged period of time. It causes injuries like alveolar damage and decrease pulmonary function, which is fatal to animals. To avoid pulmonary oxygen toxicity, small animals should not receive a FiO2 of more than 60 per cent for longer than 24 to 72 hours.

Conclusion

Success of oxygen therapy varies from patients to patients and depends mostly on the severity of the disease. As tolerance of each method varies from patient to patient, determining the most suitable method can be a challenging task, which needs experience and skilled personnel. While delivering oxygen to the patients care should be taken that oxygen administration should be stress free and if the animal becomes anxious or frightened, and starts to struggle, an alternative method should be initiated. Choosing the suitable method along with the flow rate of oxygen and careful observation of patient is the key of successful oxygen therapy.

Exercise: 5 **Date:**

GASTRIC LAVAGE

Gastric lavage is the term used when referring to a procedure that involves removing contents from the stomach. Gastric lavage in dogs is commonly called –pumping the stomach,‖ as the fluids are taken upward from the stomach organ. A gastric lavage procedure is commonly used in situations when the option of inducing emesis (vomiting) in unobtainable. Unconscious patients, ingestion of a large quantity of a toxic agent, or sometimes, in the case of gastric dilation volvulus syndrome (twisted stomach), a gastric lavage procedure is helpful. Gastric lavage procedures are often performed an emergency situation.

Gastric Lavage Procedure in Dogs

First perform a routine diagnostic examination of the canine. Routine testing will include a physical exam, blood work, a urinalysis and possibly a fecal examination. Radiographs and/or an ultrasound of the abdomen may also be taken to establish the presence of gastrointestinal obstruction or abnormality prior to conducting the gastric lavage procedure. An IV catheter will be placed, preferably in either the right or left front limbs of the dog. An intravenous catheter will allow easy access for fluid therapy and intravenously administered drugs. The canine will be given a sedative injection and will be intubated with an endotracheal tube, which will allow the veterinary team to provide the dog with oxygen and an anesthetic gas. The cuff of the endotracheal tube will be inflated to prevent gastrointestinal fluids that will be aspirated from the stomach, from entering the lungs. The patient‘s vital signs will be checked and monitored throughout the procedure. An anti-emetic drug may be administered intravenously at this time to prevent the dog from retching while the tube has been placed. The dog will be placed in sternal (on the belly) or right lateral (on the side) recumbency. The orogastric tube will be pre-measured to an appropriate size. Place the tube alongside the dog‘s body, placing the end of the tube at the last rib. The other end of the tube will then be marked with tape to ensure once the tube is inserted, it will not pass deeper into the digestive system. The tube will be lubricated and inserted into the dog‘s mouth, passing down the esophagus, then into the stomach. Then confirm that the orogastric tube is properly placed through palpation or simultaneous auscultation. Warm water will be infused down a funnel and into the orogastric tube. Using the force of gravity, the

exposed end of the orogastric tube will be directed towards a bucket on the floor for the stomach fluids to pour into. More fluids will be administered and allowed to pour out until it is felt that stomach has been lavaged. (Usually no more than ten times.) Before the tube is removed, activated charcoal will be passed through the tube. The activated charcoal will bind and –trap| any toxic substance left behind. The lavage process will once again take place to remove the captured toxins and charcoal. The orogastric tube will be kinked and removed from the dog‘s stomach. The patient will be extubated (removal of endotracheal tube) when the gag reflex returns and allowed to awaken in the recovery area.

Efficacy of Gastric Lavage in Dogs

Gastric lavage for dogs is a highly effective way to remove a toxin from the stomach before the body ingests the element.

Gastric Lavage Recovery in Dogs

After the gastric lavage procedure has been completed, the dog should show signs of improvement in the following hours. Additional motorization may be required.

Dog Gastric Lavage Considerations

The tracheal wash procedure does require the patient to undergo a brief period of anesthesia, which is the primary concern for most dog owners. Gastric lavage also poses the risk for respiratory effects (hypoxemia), mechanical injuries (mouth, throat, stomach irritation) and aspiration pneumonia if the endotracheal cuff was not properly inflated.

PERITONEAL LAVAGE

Peritoneal lavage is a diagnostic, surgical procedure performed to establish whether there is any free floating fluid (usually blood) in the abdominal cavity. A peritoneal lavage is used relatively early on, as it is primarily a diagnostic tool. Despite it being a highly accurate test for evaluating intraperitoneal hemorrhage, or ruptured hollow viscus, it is not performed frequently as abdominal sonography is increasingly being used.

Peritoneal Lavage Procedure in Dogs

Give the dog a physical examination, followed by possible tests, including blood work. Once the decision for a peritoneal lavage has been made, the procedure will take place promptly, often within days. The procedure itself will go as follows: The dog will be intravenously anesthetized. A vertical incision of the skin will be made one third of the distance from the umbilicus to the pubic symphysis. The linea alba is then divided. The peritoneum is picked up to prevent bowel perforation. The peritoneum is then entered. A catheter will be inserted towards the pelvis and an attempt at aspiration of material using a syringe will be made. If no blood is aspirated, warm saline is infused. After a few minutes this is drained and then sent for analysis.

Efficacy of Peritoneal Lavage in Dogs

Peritoneal lavage is extremely effective in attaining its goal of allowing a thorough and accurate evaluation of a hemorrhage. Due to the diagnostic nature of this procedure, plus the emphasis on safety, long-term implications are unlikely. Despite minimal risks of complications, it is extremely unlikely a dog will suffer from any permanent effects, as the procedure is used mainly for diagnostic purposes. There are alternatives to having a peritoneal lavage. Abdominal sonography is increasingly being used instead. This is due to the non-invasive nature of sonography. A peritoneal lavage is an invasive procedure, that comes with greater associated risks than a sonography. But, a sonography cannot provide as accurate results, as it does not allow direct access to the problem area. As sonography is not as informative, it increases the chance of missing vital information.

Dog Peritoneal Lavage Considerations

There are minimal risks associated with the procedure, just 0.8%-1.7% suffer from complications. Those complications are all short term issues, such as injury to the iliac vessels, inadequate fluid return, and bladder punctures. However, when the chances of these are around 1%, the benefits that the procedure offer far outweigh the risks. A peritoneal lavage can give a detailed and accurate diagnosis of the abdominal problem, which then allows for the most effective treatment to be administered. While there is the chance of the dog suffering from peritonitis and bleeding again in the future, it will not be as a result of the procedure itself.

Exercise: 6 **Date:**

COLLECTION AND EXAMINATION OF CEREBROSPINAL FLUID

Collection of CSF

For collection of CSF the material required is 12.5 cms long 14G needle with a stylet, 3 inch 16 G spinal needle, cotton swab, BP handle with knife, rectified spirit, sterilized test tubes, sharp razor etc in sterilized condition and aseptic precautions should be taken during collection.

Methods of collection

The CSF is collected from two sites,

A. Cisterna magna or atlanto-occipital puncture in horse, cat and dog.

B. Sub lumbar or lumbosacral puncture in cow, sheep and goat.

A. **Atlanto occipital puncture**: The collection can be done either in the standing position or in the lateral recumbency with following steps,

1. Restrain the animal, casting with rope.

2. Sedation of the animal by giving local anesthesia such as 2% Xylocaine, Lignocaine and tranquilization with Siquil or Largactil.

3. Flex the head by bending and stretching the neck, so that it forms a 900 angle with the longitudinal axis of the neck and hold it in this position firmly.

4. Clip, clean and sterilize the selected area.

5. Use a 3-4‖ long and 16 G spinal needle with a stylet and insert it slowly at the cranial edge of the wings of atlas. Direction of the needle should be parallel to the long axis of the head. When the needle enters subarachnoidal space resistance is not felt.

6. Needle enters into atlanto-occipital joint, remove the stylet so that the CSF flows out and collect approximately 1 to 2 ml of CSF.

B. **Sub lumbar puncture**: The collection is done in the standing position.

1. Presurgical and aseptic precautions are taken and the depression between the dorsal process of last lumbar vertebra and cephalic end of median crest of sacrum is palpated. Needle is passed and punctured at this site. Insert the needle vertically, then slightly oblique by applying gradual pressure in forward and backward directions. As the needle enters in subarachnoid space comparatively less resistance is felt.

2. Animal must be tied firmly to avoid damage to the spinal cord. CSF collection is done by removing the stylet, apply a syringe and suck the fluid.

Examination of CSF

The CSF is examined for following tests,

I. The Physical examination is described in Table.

Parameters	**Observation**	**Inference**
Colour	Clear, watery and transparent	Normal
	Red	Puncture of blood vessel during collection
	Dull red / brownish	Intracranial hemorrhage, cranium fracture
	Yellow (xanthochromic)	Presence of bile pigments (jaundice), hemorrhage.
	Grayish or greenish	Due to infection leading to pus formation
Turbidity	Clear, transparent	Normal
	Hazy, ground glass like	Presence of cells/ white

		clots appearance (pleocytosis)
	Cloudy/purulent	Encephalitis, bacterial meningitis.
	Red turbid	Puncture of blood vessel during collection
Coagulation	No coagulation	Normal
	Coagulation	Presence of abnormal amount of proteins especially fibrinogen in cases of meningitis
	Blood (in large quantities)	Internal hemorrhage or improper collection.

II. Chemical examination

a. Proteins: Normal protein content of CSF is 12-40 mg/100 ml and most of it is albumin.

b. Glucose: The quantitative estimation of CSF glucose is done by the Folin – Wu technique. The concentration of the glucose in CSF is approximately 60-70 % of blood glucose level and ranges from 40-80 mg/100 ml in normal CSF.

The glucose level in CSF depends upon the

1. Blood glucose levels,
2. Selective permeability of the blood to CSF barrier,
3. Presence or absence of glycolytic barrier.

An increased glucose level in the CSF is termed as –hyperglycorrhacia‖ and is seen in association with any disease having a hyperglycemia (Diabetes mellitus), encephalitis, spinal cord compression, brain tumors or brain abscess. A decreased glucose level in the CSF is termed as ‗hypoglycorrhacia' and is associated with systemic hypoglycemia or acute pyogenic infection.

c. Chlorides: Normal CSF values in domestic animals ranges between 650-850 mg/100ml. Lower values are seen in pyogenic meningitis, protracted vomiting, advanced pneumonia, hypochloremia, while normally higher values of chlorides in CSF are recorded than in serum.

d. Sodium: Slightly higher in CSF than in blood in salt poisoning cases.

e. Cholesterol: Hemorrhages in the CNS, tumors, meningitis and brain abscess lead to an increase in cholesterol content. Usually normal cholesterol level is very low and values recorded in Horse: 0.36 - 0.55 mg/dl and Goat: 0.51 mg/dl.

f. Determination of enzymes: Increased levels of CSF GPT: 20.1(9-46 unit) and GOT: 13.7 (2-32 units) have been observed in dogs suffering from distemper with involvement of CNS, purulent meningitis and cerebral infarction.

Lactic dehydrogenase enzyme level of CSF are also increased in bacterial meningitis, metastatic carcinoma, lymphoid tumor, subarachnoid hemorrhage and cerebral infarction. A marked elevation in the CPK (creatinine phosphokinase) is also seen in certain neurological conditions.

g. Calcium: Normally the calcium is lower in CSF than in serum. Increased level of protein bound calcium in CSF indicates disturbance in blood brain barrier.

Table. Chemical Examination of CSF:

Tests	**Observation**	**Inference**
Foam test Take CSF in test tube and shake the test tube at least for 5 minutes	Slight foam that disappears after few minutes More foam that remains Protein level Increased	Normal Protein levels
Sulfosalicylic acid test (SSA) 3 ml of 3% SSA + 1 ml CSF Mix and allow to Stand	Increase in the turbidity	Presence of proteins
Nonne - Apelt test 1 ml saturated ammonia solution + 1 ml CSF Do not mix. Allow to stand	White to grayish ring at the junction of two fluids	Presence of increased amounts of globulin in CSF which is seen in 1. Encephalitis, 2. Meningitis, 3. Neoplasia, 4.Hemorrhage, 5.Hydrocephalus, 6.Tissue destruction, 7. Uremia, 8.Toxoplasmosis, 9. Pneumonia
Pandy's test 1 ml saturated phenol or Pandy's reagent, 1-2 drops of CSF Shake (reagent is prepared by dissolving 10 grams of pure phenol in 150 ml of distilled water)	White cloudy or turbid	

III. Cytological examination

The total cell counts of the CSF must be estimated within 20 minutes of collection, since the cells degenerate rapidly. The estimation of the number of cells is done as for the determination of WBC‘s of the blood. The total number of cells which are obtained are then multiplied by 0.6 to get number of cells in one cu mm of the CSF.

Normal counts:

Cattle, sheep and pig 0 - 15 cells/ cu mm

Dog upto 25 cells/cu mm

Horse upto 23 cells/cu mm

Pleocytosis or increased number of WBC‘s are seen in inflammatory conditions of brain, spinal cord or meninges, abscess of brain or spinal cord, encephalitis, chronic inflammatory conditions, toxic or degenerative conditions.

Differential Count: Prepare the smear from CSF, dry it and stain with leishmanns stain, examine under microscope. Neutrophilia indicates pyogenic or bacterial infection, abscesses in brain, bacterial meningitis, encephalitis and hemorrhage, while lymphocytosis is observed in uremia, toxemia, chronic viral and fungal infection.

IV. Bacteriological examination

It is carried out when the CSF cell count and protein contents are high. The organisms are isolated in CSF and identified by cultural methods.

Exercise: 7 **Date:**

BLOOD TRANSFUSION

Blood transfusion is being practiced for centuries for saving life of human beings and animals. Richard Lower in 1665 transfused the blood in a dog for the first time in the history. With the help of latest techniques and equipment developed after 1950, blood transfusion became more popular in veterinary medicine. Blood transfusion has made considerable advancements in veterinary medicine in recent times. Although, the information and availability of blood and its products has increased, transfusion therapy has become more complex. Advanced screening facilities, blood group testing and techniques for cross matching blood had made the process of donor selection more complicated. Advancement in techniques of separating the components of blood has given the clinician an opportunity to use the component as per the demand of the patient.

Blood groups in various animals

Animals	**Blood groups**
Dog	6
Cat	3
Horse and donkey	7
Cattle	11
Sheep and goat	7

Dog

The blood group system in dogs includes DEA 1.1, DEA 1.2, DEA 3, DEA 4, DEA 5 and DEA 7. DEA 1.1 and 1.2 are the most important blood groups and are found in 60% population of canines. 15 ml of blood per kg BW can be collected from dog in every 6 weeks.

Cat

Three blood groups are reported in cats in AB blood group system. Type A blood group is the most common group and found in 95% of the American cats. Majority of Indian and 30 % of cats in UK belongs to blood group B. Blood group AB is extremely rare. Healthy adult cats can donate 45-60mL every 6 weeks.

Horse and donkey

The seven blood groups in horses viz. A, C, D, K, P, Q and U are internationally recognized with more than 30 erythrocyte antigens. Universal donor horse is not possible because of various possible antigenic combinations. The cross matching must be performed although impractical to minimize transfusion reactions. Adult horses can safely donate approximately 6-8 L of blood. Whole blood can be collected every 15-30 days and plasma collected every 7 days if the erythrocytes are returned to the donor.

Cattle

The internationally recognized blood groups in cattle are A, B, C, F, J, L, M, R, S, T and Z. Out of these 11 groups, group B and J being the most clinically relevant. Cattle can donate 8-14 mL of blood per kg of body weight.

Sheep and goat A, B, C, D, M, R, X are the seven blood groups identified in sheep.

Cross-matching technique

- Collect the blood from the donor as well as recipient in purple top and red top tubes i.e EDTA tube and non-EDTA tubes respectively.
- Centrifuge the blood and allow separating plasma and serum from the RBCs.
- Remove the serum and save it in a separate sterile tube.
- Discard the plasma from the EDTA tube.
- Wash the RBCs collected from EDTA tube.
- Place the RBCs in a spate tube filled with normal saline and centrifuge for 1 minute.
- Repeat the process 5 times removing the supernatant every time.
- Resuspend the cells to make a 2% to 4% solution (0.2mL of blood in 4.8mL of saline gives a 4% solution).

- Label the tubes to make the following mixtures as Major crossmatch (2 drops patient serum with 1 drop donor RBC suspension), Minor crossmatch (1 drop patient RBC suspension with 2 drops donor serum) and Control (1 drop patient RBC suspension with 1 drop patient serum). Incubate the mixtures for 15 to 30minutes at 37°C and then centrifuge for 15seconds.
- If either hemolysis or hemagglutination is seen macroscopically, or if agglutination is seen microscopically, the donor is not a good match.

Transfusion therapy: General principles and indications

- Blood transfusion should be practiced after proper blood grouping and cross matching the donor‘s group with the recipient‘s to prevent the transfusion reactions.
- In addition to potential adverse reaction of mismatched blood transfusion, the shortened lifespan of mismatched transfused cells result in ineffective therapy.
- Breeding females of all the species should be properly checked for bold group and cross matched to dodge primary sensitization and risk of future offspring developing hemolytic disease. In the past, cross matching was recommended in dogs that had previously been pregnant. However, a recent study showed that pregnancy does not seem to sensitize dogs to antigens on RBCs.42 Blood grouping for canine DEA 1.1 and for feline types A and B generally practiced in veterinary medicine. Other groups and cross matching can be done in research and reference laboratories.
- Blood transfusions are mostly risky, hence, they should be performed in only warranted cases.
- History of previous transfusion therapy should be collected from clients, which necessitates cross matching. Whole blood as well as component is transfused in veterinary medicine depending upon availability and indications of transfusion.
- The primary indication for blood transfusion is the treatment of severe anemia caused by hemorrhage, hemolysis, ineffective erythropoiesis, immune-mediated hemolytic anemia, chronic inflammatory or infectious disease, or neoplasia. Animals must be clinically evaluated on an individual basis.
- A thumb rule for the treatment of anemia is to transfuse when the packed cell volume (PCV) is less than 10% to 15%. Animals with acute-onset anemia, however, usually

require transfusion before their PCV decreases to 15%, which contrasts with the situation in animals with chronic anemia.

- For cases of thrombocytopenia, the generally accepted trigger for platelet transfusion is platelet counts of 10,000/μL.
- Additional indications for transfusion include hypovolemia, primary or secondary clotting factor deficiency and hypoproteinemia.
- Collected blood should be labeled with all the details and record keeping is crucial in all cases of blood collection and administration.

Selection of donor

- Blood grouping should be performed to select permanent blood donors. All donors should be healthy young adults that have never been transfused.
- In addition, donors must have undergone routine physical, hematological and clinical chemistry evaluations examinations.
- Proper clinical history of the expected donor should be collected by carefully interviewing the owner to minimize the risk of disease transmission through blood.
- In the veterinary medicine, it is usually the cost that restricts to test individual units. Therefore, a combination of careful interview and blood screening of the donor is used to minimize the risk of infectious disease transmission.
- Donor should be properly vaccinated and should be tested free of blood parasites and other infectious diseases.
- Donors should have normal baseline PCV and total protein concentrations prior to any donation. Blood should be collected aseptically usually via jugular venipuncture. To avoid interference with platelet function, donors should not be sedated with acepromazine.

Anticoagulants used for storage

Two anticoagulants namely Citrate-phosphate-dextrose-adenine (CPDA-1) and Acid-citrate-dextrose (ACD) are commonly used for storage of blood. CPDA-1 is considered better anticoagulant because it maintains higher levels of 2,3-disphosphoglycerate (2,3-DPG) and adenosine triphosphate (ATP) in collected blood. In CPDA-1 blood can be stored for

approximately 35days. Acid-citrate-dextrose (ACD) allows storage of blood for 21 days. Use 1mL of anticoagulant (CPDA-1/ACD) for every 7mL of blood. Blood should be refrigerated in plastic blood collection bags. Heparin is not recommended for blood collection because it activates platelets, but if still used 5 U per mL of blood is sufficient. Blood collected in heparin as anticoagulant must be used immediately. Survival and functional usefulness of erythrocytes decrease with increased storage temperature and time because of glucose consumption and depletion of ATP and 2,3- DPG. Blood should be collected into latex free plastic bags or plastic syringes to preserve platelets.

Transfusion process

Aseptic conditions should be maintained and perfect aseptic procedures should be followed while collection of blood for transfusion. A separate aliquot from every donated unit of blood should be stored for later testing when disease transmission following transfusion is suspected. Blood should be filtered using 150-170μm pore nonlatex filters either prior to or during administration. Blood should be warmed to 37°C before administration to prevent hypothermia. Temperature should not be more than 37°C, because higher temperatures cause lysis of erythrocytes and inactivation of clotting factors. Blood is administered intravenously through commercially available administration I/V sets with filters. Fluid containing 0.9% saline should be used when concurrent crystalloid fluid therapy is indicated or for reconstituting blood components such as packed erythrocytes. Lactated Ringer‘s solution causes calcium chelation with citrate-containing anticoagulants and subsequent clot formation, 5% dextrose in water cause swelling and lysis of erythrocytes and hypotonic saline fluids will lyse erythrocytes, so they are contraindicated. Excessive and rapid injection of blood or plasma can result in circulatory overload and heart failure. Generally, blood should be given intravenously at a rate not exceeding 10mL/kg per hour (always begin the transfusion slowly then gradually increase flow rate); however, each patient must be assessed individually to establish an appropriate infusion rate. For example, hypovolemic patients may require an infusion rate of 20mL/kg per hour, whereas patients with cardiac, renal, or hepatic disease or recumbent calves may require an infusion rate of only 1mL/kg per hour. If blood is transfused too quickly, salivation, vomiting and muscle fasciculations may occur. Warm blood should be transfused within 4 hours to avoid contamination. The volume of blood to be transfused is determined according to the recipient‘s

body weight, estimated blood volume, PCV of the recipient and of the donor and the purpose of therapy. A simple guideline for small animals is that 10-15mL/kg of packed erythrocytes or 20mL/kg of whole blood increases the PCV by 10% if the donor has a PCV of approximately 40%. One report in horses demonstrated that 15mL/ kg of whole blood and 8-10mL/kg of packed erythrocytes resulted in a 4% increase in PCV when the donor PCV was 35-40%. More specific calculations for cattle are reported depending on the indication for transfusion for example, in hemorrhagic shock a general volume rule is 7L of whole blood/600kg cow.21 For cases of thrombocytopenia in dogs and cats for which fresh whole blood is used for treatment, the general rule is to administer 10mL/kg to increase the recipient‘s platelet count by a maximum of about 10,000/µL. In dogs, the half-life of erythrocytes transfused after matching is approximately 21days. In cats, the half-life of erythrocytes transfused after matching is approximately 30- 38days. In horses and cattle the survival time of compatible transfused erythrocytes is only 2-4 days.

Initial Infusion Rate

0.25 mL per kilogram (kg) of body weight over a 30 minute period

Caution: Dog should be watched carefully during this process for any allergic reactions

Infusion Rate

If no problems arise in the receiving patient after the initial 30 minutes, the rate may be increased.

The recommended infusion rate is 22 mL/kg per day.

Total Blood Volume to be infused

Total mL donor dog blood in anticoagulant = 2.2 x receiving dog‘s weight (kg) x 40 x packed cell volume (PCV) desired in receiving dog - PCV of receiving dog / PCV of donor dog blood in anticoagulant.

Note: 2.2 mL/kg of whole blood will raise PCV by 1% when PCV of transfused blood is 40%.