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INTRODUCTION


The purpose of this document is to provide research investigators and members of Animal Ethics Committees with information that will assist in evaluation of blood collection procedures proposed for a selection of laboratory animal species. It is intended as a brief summary, however additional information can be obtained from the document prepared by the BVA/FRAME/RSPCA/UFAW Joint Working Group on Refinement (see References). Blood collection is a procedure that is frequently performed in the course of animal experimentation. The procedure impacts upon the pain and distress experienced by the animal, therefore Refinements to the methodology must be considered in order to meet the requirements of the *Australian Code of Practice for the care and use of animals for scientific purposes* (7th Edition, 2004). There are differences in the methodology selected according to the species, individual animal characteristics (eg. age, sex), the volume of blood which is required, and the requirements of the experiment. **A number of recommended methods are therefore suggested, however this does not preclude use of other methods, which may be justified on a case-by-case basis.** The guidelines apply to Mice, Guinea Pigs, Rabbits, Pigs, Cats, Chickens, Marmosets and Macaques, Amphians, Reptiles and Fish. For other livestock species namely Cattle, Sheep, Horses and Goats please see "Guidelines for the harvesting of Blood from Livestock for Commercial and Research Purposes 2007" by Bureau of Animal Welfare, Department of Primary Industries, Victoria on: <http://www.monash.edu.au/research/ethics/animal/regguide/bloodharvesting0907.pdf>

RECOMMENDED MAXIMUM VOLUME OF BLOOD COLLECTION IN LABORATORY MAMMALS

- These recommended maximum volumes relate to normal, healthy animals on a satisfactory plane of nutrition.
- Collection of blood from animals which are sick, dehydrated or heat stressed is not recommended as the additional stresses associated with the procedure would further compromise animal health.
- Fresh drinking water must be available to the animal to enable it to regain normal fluid balance.

Single Blood Sample

- The recommended maximum volume of blood collected as a single sample is 10% of the circulating blood volume*.
- If greater volumes are required to be withdrawn, then the animal should receive fluid replacement equal to the volume collected, and be monitored for signs of haemorrhagic shock (rapid heart rate, pale mucous membranes, cold skin and extremities, increased respiration rate). The fluid replacement recommended is warm (37 °C) sterile isotonic physiological solutions such as Normal (0.9%) Saline, or Hartmann's Solution, or Ringer's Lactate. It can be given by subcutaneous injection, or slow intravenous injection.
- Removal of 30% (or greater) of the circulating blood volume is hazardous to the animal's health, as there is a significant risk of shock and death.
- A single sample blood collection can be performed every two weeks, however in this situation haematocrit (packed cell volume) and other haematological assessments must be made regularly, as there is an increased risk of anaemia developing over time. (Refer Appendix)
- For example: To estimate a safe volume of blood that can be collected weekly from a 25g mouse (assume a milliliter of blood weighs 1.0g). $25g \times 79ml/1000g = 25g \times 0.079ml/g = 1.98 \text{ ml total blood volume}$ $1.98 \text{ ml} \times 10\% \sim 0.2 \text{ ml of blood (200}\mu\text{l)}$

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Repeated, Frequent Blood Sampling

- The recommended maximum volume of blood collected for repeat bleeds at intervals less than 2 weeks is: 1% of the circulating blood volume* every 24 hours, or 7% per week.
- Haematological assessments must be made regularly, as there is an increased risk of anaemia developing over time. (Refer Appendix)
- Consider placement of an indwelling venous cannula if many samples of blood are to be collected over a short timespan (hours or a few days). This will reduce the number of venepunctures, and may enable collection of blood with minimal animal stress.

*As a general guide, the blood volume of most animals is approximately 6% of the animal's bodyweight (Mc Guill and Rowan, 1989) or in the range 55-70 mL/kg (Joint Working Group on Refinement, 1993). Refer to the table in the Appendix for species-specific information.

RECOMMENDED SITES AND EQUIPMENT FOR COLLECTION OF BLOOD IN MAMMALS

- These sites and methods are commonly used, and are recommended for reasons of ease of blood collection and minimisation of animal stress. In each case, the appropriate receptacle for the blood and other appropriate materials (such as anticoagulant for plasma collection) are also required.
- Needle size is a guide only. Smaller needles than those indicated may be needed for very young animals. Use as large a diameter (lowest gauge) needle as possible to enable rapid sampling with minimum haemolysis.


MOUSE

R =Recommended; A = Acceptable with reservations

	Sites	Comments	Equipment
R	Tail vein puncture or section	Anaesthesia recommended. Warm animal. Small volume (a few drops)	26-30G needle, capillary tube in needle hub; lancet
R	<u>Submandibular Bleeding</u>	A simple way to obtain blood by puncturing the area behind the hinges of the jawbones	mouse bleeding lancets
R	Saphenous vein	Anaesthesia recommended. Small volume (a few drops)	23G needle or lancet; to pierce vein; capillary tube or syringe or pipette to aspirate drops.
R	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume). Clean sample.	25G needle 1 or 2 mL syringe
A	Orbital venous sinus*	Anaesthesia required. Up to 0.25mL blood.	Capillary (microhaematocrit) tube to puncture sinus and collect blood
A	Tail tip amputation**	Anaesthesia required. Maximum twice. Warm animal. Small volume (a few drops)	Scalpel blade to amputate 2-3 mm of tail tip. "milk" tail to collect blood.

* The use of this method has become controversial due to concerns that eye damage or infection may sometimes occur. If performing more than once, alternate between the two orbits.

** This method has become controversial as loss of the tail tip is unaesthetic, and there is increased risk of infection and pain if bone is sectioned. Cutting the tail too short may result in trauma to the cartilage and ultimately to the coccygeal vertebrae

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RAT R =Recommended; A = Acceptable with reservations


	Sites	Comments	Equipment
R	Tail vein puncture or section	Anaesthesia recommended. Warm animal.. Blood obtained using needle & syringe, or by nicking/puncture of vein. Moderate volume (0.5-1.5 mL)	23-26G needle; 1 mL syringe; or ; lancet or small scalpel blade and capillary tube. tourniquet to raise vein
R	Saphenous vein	Anaesthesia recommended. Small volume (approx 0.5 mL)	25G needle or lancet to pierce vein; capillary tube or syringe or pipette to aspirate drops.
R	Jugular vein	Anaesthesia and skin incision to visualise vein recommended. Moderate volume. Clean sample.	21G needle; 2 mL syringe Skin closed using suture or Micheal clip.
R	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume). Clean sample.	23G needle 5 or 10 mL syringe
R	Caudal Vena Cava	Terminal bleed only. Surgical (deep) level of anaesthesia required. Largest volume collected (approx. half blood volume). Clean sample.	Surgical instruments to open abdomen. 21-23G needle 5 or 10 mL syringe
R	Dorsal Aorta	Terminal bleed only. Surgical (deep) level of anaesthesia required. Largest volume collected (approx. half blood volume). Clean sample.	Surgical instruments to open abdomen. 23G needle and 5 or 10 mL syringe
R	Axillary vessels	Terminal bleed only. Euthanasia required just prior to collection. Large volume collected (approx. half blood volume). Sample may be contaminated with hair and tissue fluids.	Use scalpel blade to severe skin and vessels in armpit. Aspirate blood using a pipette, or alternatively use a 21G needle and 5 or 10 mL syringe.
A	Tail tip amputation*	Anaesthesia required. Maximum twice. Warm animal. Small volume (up to 0.5 mL). Haemostasis required	Scalpel blade to amputate 0.5cm tip. "milk" tail to collect. gelatine sponge for haemostasis..

R =Recommended; A = acceptable with reservations

* This method has become controversial as loss of the tail tip is unaesthetic, and there is increased risk of infection and pain if bone is sectioned. Removal of skin and subcutaneous tissue only (not bone) is acceptable.

GUINEA PIG R =Recommended; A = Acceptable with reservations

	Sites	Comments	Equipment
R	Cardiac	Recovery and terminal bleeds. Anaesthesia required. Largest volume collected (approx. half blood volume).	23-21G needle 5 or 10 mL syringe
R	Saphenous vein	Anaesthesia recommended. Small volume (approx 0.5 mL)	25G needle or lancet to pierce vein; capillary tube or syringe or pipette to aspirate drops.
R	Anterior vena cava	Anaesthesia required. Moderate volume (0.5-1.5 mL).	23-21G needle; 2 mL syringe
A	Ear vein	Warm animal. Small volume (a few drops)	27G needle or lancet to pierce vein; capillary tube or syringe or pipette to aspirate drops.
A	Jugular vein	Anaesthesia and skin incision to visualise vein recommended. Moderate volume. Clean sample.	23G needle; 2 mL syringe Skin closed using sutures.

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RABBIT R =Recommended; A = Acceptable with reservations


	Sites	Comments	Equipment
R	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume).	21G needle 20 mL syringe
R	Marginal ear vein	Warm animal and ear. Recommend use of topical anaesthetic spray or gel to blood vessel site. Moderate volume collected (5-10 mL)	23G needle and 5-10 mL syringe; or lancet to pierce vein then collect drops
R	Central Ear Artery	Warm animal and ear. Recommend use of topical anaesthetic spray or gel to blood vessel site. Moderate volume collected (5-10 mL)	23G needle and 5-10 mL syringe.
A	Jugular vein	Percutaneous blood collection possible by skilled operator, however anaesthesia and skin incision to visualise vein recommended. Moderate volume. Clean sample.	23G needle; 2-5 mL syringe Skin closed using sutures.

PIG R =Recommended; A = Acceptable with reservations

	Sites	Comments	Equipment
R	Cranial Vena Cava	Suitable for young pigs; physically restrained on back; or mature pigs physically restrained with pig snare, standing	50 mm long 19-21G needle and syringe or vacutainer; 75 mm long 14G needle (free flowing)
A	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume).	8-10G needle and tubing
A	Ear vein	Recommend sedation and confinement in small pen. Small volume (up to 5 mL).	21G butterfly needle and 5 mL syringe; or lancet and collect drops
A	Femoral vein	Anaesthesia and skin incision to visualise vein recommended. Moderate volume. Clean sample.	23G needle; 5 or 10 mL syringe Skin closed using sutures.
A	Jugular vein	Moderate volume (10 mL). Acceptable for large adults. Anaesthesia and skin incision to visualise vein recommended. Percutaneous venepuncture possible for skilled operators.	18-21G needle and 10 or 20 mL syringe or vacutainer. Sutures for skin incision.

CAT R =Recommended; A = Acceptable with reservations

	Sites	Comments	Equipment
R	Cephalic vein	Sedation recommended for temperamental animals. Moderate volume collected (approx. 5 mL).	23-21G needle 5 or 10 mL syringe
R	Jugular vein	Sedation recommended for temperamental animals. Percutaneous venepuncture. Moderate volume (5-10 mL).	21G needle; 10 mL syringe
A	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume).	21G needle and 10 or 20 mL syringe.
A	Femoral vein	Moderate volume. Clean sample.	23G needle; 5 or 10 mL syringe

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CHICKEN **R =Recommended; A = Acceptable with reservations**

	Sites	Comments	Equipment
R	Wing (brachial) vein	Moderate volume collected (up to 5 mL).	25-23G needle (bent); 5 mL syringe
A	Anterior vena cava	Anaesthesia required. Moderate-large blood collection (approx.10-20 mL)	Long (70mm) 23G needle; 10 mL syringe
A	Jugular vein	Moderate volume (5mL).	23G needle; 5 mL syringe
A	Wattles, comb	Small volume (a few drops)	Lancet to prick; collect drops.
A	Cardiac	Terminal bleed only. Anaesthesia required. Largest volume collected (approx. half blood volume).	23-21G needle and syringe

MARMOSET **R =Recommended; A = Acceptable with reservations**


	Sites	Comments	Equipment
R	Femoral vein	Moderate volume (1-3mL).	25G needle; 1 mL syringe
A	Jugular vein	Anaesthesia required Moderate volume (1-3mL).	25-23G needle; 1-2 mL syringe
A	Saphenous vein	Small volume.	23G needle or lancet to pierce vein; capillary tube or syringe or pipette to aspirate drops.

MACAQUE **R =Recommended; A = Acceptable with reservations**

	Sites	Comments	Equipment
R	Femoral vein	Anaesthesia required. Moderate volume (10-30mL).	23G needle; 10 mL syringe
R	Cephalic vein	Small volume (a few mL).	23G needle and syringe.
A	Jugular vein	Anaesthesia required. Moderate volume (10-30mL).	23-21G needle; 10 mL syringe
A	Saphenous vein	Small volume.	23G needle or lancet to pierce vein; capillary tube or syringe or pipette to aspirate drops.

REFINEMENTS TO MINIMISE ANIMAL STRESS

- Blood collection should only be performed by suitably trained personnel. Seek training and advice if an unfamiliar technique is to be performed.
- Blood collection from animals which are sick or physiologically abnormal is not recommended.
- Blood collection can be distressing to the animal being handled. Distress vocalisations, blood, excreta and pheromones from the animal may cause distress to other animals in the same environment, and also to human observers. As a general principle animal procedures should be performed in an area separate from other animals.
- Animals must be adequately restrained in order to minimise risk of injury to both the animal and operator. The method of restraint selected (physical, chemical, or combination) must be appropriate to the species, operator skill, and experimental requirements. Many methods of physical restraint require an experienced animal handler.
- Use of local anaesthetic skin creams in species such as the rabbit, may reduce pain associated with central artery or marginal ear vein puncture. To be effective, it must be applied 30-60 minutes before the procedure.
- Use of heat is recommended to assist with vasodilation in rats, mice and rabbits. The heat source can be an infrared lamp, or a thermostatically-controlled heating box (30°C) for whole body warming, or warm water

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(45°C) for tail warming. The animal must be continuously supervised throughout this process in order to avoid overheating (hyperthermia). Signs of overheating include increased salivation, agitation and rapid breathing.

- Use of skin irritants such as xylene (xylol) to dilate vessels is not recommended.
- Prepare the venepuncture site by clipping hair and swabbing with 70% alcohol. This will enable the vein to be more easily visualised, and will clean fat and dirt from the skin surface. Skin is almost impossible to sterilise, therefore shaving and excessive application of soaps and disinfectants are generally unwarranted, and may irritate the skin.
- At the end of the blood collection procedure, apply gentle pressure to the site for 30 seconds or longer. This will minimise subcutaneous haemorrhage

RECOMMENDED MAXIMUM VOLUME OF BLOOD COLLECTION IN AMPHIBIANS, REPTILES AND FISH

The total amount of blood that can be safely withdrawn from a reptile, amphibian and Fish depends upon the animal's size and health status. As a general guideline, blood volume is 5-8% of body weight in Oreptiles and amphibians, 1% of the body weight may be withdrawn for blood sampling, recommended volume for blood collection is 0.5-0.8 % of bodyweight . During a single collection from a non-compromised, healthy fish, blood can be collected in a volume less than or equal to 0.5 % of body weight.

RECOMMENDED SITES AND EQUIPMENT FOR COLLECTION OF BLOOD IN AMPHIBIANS, REPTILES AND FISH

These sites and methods are commonly used, and are recommended for reasons of ease of blood collection and minimisation of animal stress. In each case, the appropriate receptacle for the blood and other appropriate materials (such as anticoagulant for plasma collection) are also required. Needle size is a guide only. Smaller needles than those indicated may be needed for very young animals. Use as large a diameter (lowest gauge) needle as possible to enable rapid sampling with minimum haemolysis.


AMPHIBIANS - ANURANS (FROGS AND TOADS)

Several sites can be used depending upon size and species being sampled (Wright, 2001).

- Blood samples can be collected from the ventral abdominal vein, ventral coccygeal vein, and lingual lingual venous plexus.
- The ventral abdominal vein is often readily visible in larger amphibians and the blood collection techniques are similar to those described for lizards.
- The ventral coccygeal vein can be used in larger salamanders and newts.
- The approach to this vein is similar to that described in lizards.
- The lingual plexus, which is a complex of blood vessels located under the fleshy tongue, can be used for larger amphibians. A soft, rubber speculum should be used to gently open the animal's mouth. A cotton tipped applicator should be used to displace the tongue and facilitate exposure to the venous plexus. A needle (with no syringe attached) can be inserted into the plexus and blood can be collected out of the hob of the needle using a microhematocrit tube.
- Blood may also be collected using cardiac puncture; however, this is an invasive procedure and should be carried out under anaesthesia unless the animal is adequately restrained.

CHELONIANS (TURTLES, TERRAPINS AND TORTOISES)

Several sites can be used in obtaining blood from chelonians, each having advantages and disadvantages. Sites include jugular vein, brachial vein, ventral coccygeal vein, subcarapacial vein (Gandal, 1958; Dessauer, 1970; McDonald, 1976; Maxwell, 1979; Taylor and Jacobson, 1981; Roskopf, 1982; Jacobson, 1987).

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- Blood samples can be collected from the jugular vein, dorsal coccygeal sinus, the brachial or vein Subcarapacial vein.
- Blood collection in chelonians may require sedation.
- The right jugular vein is generally more prominent than the left and is located on the lateral aspect of the neck, approximately at the level of the ear (tympanum). An assistant should gently retract the head to facilitate exposure. An index finger should be placed on the neck to assist with jugular vein visualization.
- The brachial vein is located on the posterior aspect of the elbow. A 22-25 gauge needle should be inserted perpendicular to the posterior aspect of the elbow and negative pressure applied immediately after penetrating the skin.
- The dorsal coccygeal sinus is located on the dorsal midline of the tail. The tail should be extended and a 25-gauge needle inserted on the dorsal midline in a cranial direction.

CROCODILIANS (CROCODILES)

- Blood samples can be obtained ventral coccygeal, Ventral abdominal or Supravertebral veins or heart via cardiocentesis (Jacobson, 1984).
- Supravertebral vessel located caudal to the occiput and immediately dorsal to the spinal cord (Olson et al., 1975). A 3.75-cm, 22- or 23-gauge needle is inserted through the skin in the midline directly behind the occiput and is slowly advanced in a perpendicular direction. As the needle is advanced, gentle pressure is placed on the plunger. If the needle is passed too deep, the spinal cord will be pithed.
- Other sites of blood collection that are commonly used include the heart (via cardiocentesis) and ventral coccygeal vein (Jacobson, 1984). The heart is located in the ventral midline, approximately 11 scale rows behind the forelimbs. In collecting blood from the coccygeal vein, the crocodilian is placed in dorsal recumbency and the needle is inserted through the skin toward the caudal vertebrae.


LIZARDS

Blood samples can be obtained from Ventral coccygeal, Ventral abdominal and Jugular Vein or cardiac puncturing cardiocentesis ((Esra et al., 1975)

- Samples are routinely collected from ventral coccygeal vein (ventral tail vein - located on the ventral midline of the tail) or the ventral abdominal vein.
- If drawing from the tail vein the sample should be collected from within the proximal 1/4 of the tail. A 22-25 gauge needle should be inserted perpendicular to the skin and inserted to the point of the caudal vertebrae. Gently apply negative pressure as the needle-syringe is withdrawn off of the caudal vertebrae.
- If drawing from the ventral abdominal vein (located on the ventral midline within the body cavity) a 22-25 gauge needle should be inserted, bevel up, at a 15-30° angle, practically parallel to the body wall. Negative pressure should be applied one the needle is inserted.
- Microcapillary tubes also can be used to obtain blood samples from the orbital sinus (LaPointe and Jacobson, 1974), in a similar fashion for collecting blood from mice.

SNAKES

The total blood volume of reptiles varies between species but as a generalization is approximately 5 to 8% of total body weight. Reptiles can tolerate 1% blood loss, recommended volume for blood collection is 0.5-0.8 ml/100 g (Lillywhite and Smits, 1984; Smits and Kozubowski, 1985). Blood samples can be obtained from a variety of sites, including the palatine veins, ventral tail vein, and via cardiocentesis (Olson et al., 1975; Samour et al., 1984).


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- The ventral coccygeal vein is located on the ventral midline of the tail, the sample should be collected in the proximal 1/3 of the tail. Collection of blood from this site is usually possible only larger snakes. The individual collecting the sample should grasp the tail. The snake's body should be elevated vertically to ensure blood flow toward the tail. A 22-25 gauge needle should be gently inserted at a 45° angle between the ventral scutes of the tail at a location between one-half and two-thirds of the distance from the cloaca to the tip of the tail and advanced to the caudal vertebrae. Inserting the needle as close as possible to the midline of the ventral surface of the tail increases the chance of hitting the centrally-located caudal vein or artery. The needle should be inserted slowly until a slight resistance from the caudal vertebrae is felt; the needle should then be withdrawn slightly (i.e. 0.5 to 2.0 mm), and then the plunger of the syringe gently drawn back.
- Cardiac puncture can also be utilised if large volumes of blood if necessary. The procedure is safe and effective as long as the heart is not excessively traumatized with multiple attempts at sampling. This method should be limited to those snakes over 300 grams (Jackson, 1981). The heart is located approximately 1/3 to 1/4 the distance from the head. The animal should be positioned on its dorsum and the heart localized by visual inspection or palpation. A 22-25 gauge needle should be inserted under the scale at the most distal point of the beating heart (ventricle). Apply negative pressure once the needle is inserted.
- The palatine vessels are located on the dorsal palate inside the mouth. The oral cavity should be opened using an appropriate speculum. The palatine vessels are located between the palatine and maxillary teeth. A 25-26 gauge needle should be inserted into the blood vessel at a 45° angle and negative pressure applied immediately. This location should only be used as a last resort.

FISH

The volume of blood/unit of body weight is considerably less in bony fish compared to mammals. Thus the recommendation for calculating the volume of blood that can be taken during a single blood collection based on a percentage of body weight is reduced. During a single collection from a non-compromised, healthy fish, blood can be collected in a volume less than or equal to 0.5 % of body weight. All fish should be anaesthetized prior to blood sampling. Blood may be sampled following four separate procedures: dorsal aorta, cardiac puncture, caudal vein, and caudal severance. Of the three sampling methods the caudal vein is the preferred method for fish. Use of 22 Ga. hypodermic needles are recommended for larger fish. Smaller gauge needles (upwards of 22 Ga.) Are recommended for smaller fish.

- Caudal vein is the preferred site for blood sampling. The vessels running beneath the vertebrae of the fish can be sampled by using a lateral or ventral approach. The sample is taken midline just posterior of the anal fin. Insert the needle into the musculature perpendicular to the ventral surface of the fish until the spine is reached or blood enters the syringe. If contact with the spine is made withdraw the needle slightly. The vein is ventral to the overlying spine. This blood vessel can also be sampled laterally.
- Dorsal Aorta: Insert needle on a 30-40° angle into the dorsal midline in the roof of the mouth at about the 3rd to 4th gill arch. Depending upon size and species of fish insertion between the 1st and 2nd arch may be more suitable. Recovering fish tend to bleed at the mouth. This site may be used for indwelling catheterization.
- Cardiac Puncture: Cardiac punctures from the ventral side are sometimes used in fusiform fish or through the operculum in laterally compressed species. Blood is collected from the heart ventricle. Insert needle perpendicular to the ventral surface of the fish in the centre of an imaginary line between the anterior most part of the base of the pectoral fins.
- Caudal Severance: Blood from the caudal vessels may be collected directly into collection tubes by cutting off the tails of anesthetized fish that will be euthanized following the procedure.. Dry the caudal peduncle. Completely sever the tail posterior to the anal fin. The first few drops are discarded, the rest is collected in microhematocrit tubules. After the sample is collected return the fish to a separate container of anaesthetic for euthanasia.

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APPENDIX: HAEMATOLOGICAL ASSESSMENT

Collection of a large volume of blood, (or small volumes on a frequent basis), may reduce the number of circulating red blood cells, and reduce the oxygen-carrying capacity of the blood. Although there are limited stores of red blood cells in the spleen which can be used to rapidly replace losses, replacement in the longer term requires production by the bone marrow. This production in turn requires an adequate supply of nutrients to the bone marrow, including iron. Where the rate of replacement of red blood cells cannot balance the rate of depletion, anaemia may occur. The clinical signs of anaemia include:

- pale mucous membranes (tongue, gums and ears)
- increased respiratory rate at rest
- exercise intolerance (easily exhausted)

Examination of blood by a veterinary laboratory will also enable detection of other signs such as:

- reduced numbers of circulating red blood cells (red cell count)
- reduced proportion of red blood cells in blood (haematocrit %; packed cell volume PCV%)
- reduced haemoglobin in blood
- presence of increased numbers of immature cells (reticulocytes)
- presence of abnormal red blood cells

The following table provides data on normal haematological measurements, for use as a guide only. **As differences exist between animal strains and between normal values obtained by different veterinary laboratories, control animal data should be collected and compared with the animal data being examined.**


Table: Normal Haematological values (Source: Joint Working Group on Refinement, 1993)

Species	Packed Cell Volume (%)	Red blood cell nos. (10 ¹² /L)	Haemoglobin (g/100 mL)	Reticulocytes (%RBC)	White blood cell nos. (10 ⁹ /L)	Blood volume (mL/kg)
mouse	35-45	7.7-12.5	10-20	3.3-13.3	8.0	78-80
rat	35-45	7.2-9.6	12-18	1.7-21.1	14.0	50-70
guinea pig	35-42	4.5-7.0	11-17	1.8-6.1	10.0	67-92
rabbit	30-50	4.5-7.0	8-15	2.9-8.0	9.0	44-70
pig	30-50	5.0-9.0	10-16	-	7.0-20.0	65
cat	30-50	6.0-10.0	8-14	0-1.0	5.5-19.5	47-66
chicken	23-55	1.25-4.5	7.0-18.6	-	9-31	60
marmoset	40-55	5.3-8.1	12-19	1.0-10.0	3.0-13.0	-(82*)

* Richter *et al.* (1984) p362

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Title	BLOOD COLLECTION GUIDELINES	

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