

University Animal Care Committee Standard Operating Procedure		
Document No: 7.10.5	Subject: Cardiac Blood Collection in Mice	
Date Issued: March 14, 2012	Revision: 5	Page No: 1

Location: Queen's University

Responsibility: Principal Investigators, Research Staff, Veterinary Staff

Purpose: The purpose of this Standard Operating Procedure (SOP) is to describe the methods of cardiac blood collection in mice.

1. Introduction and Definitions:

Abbreviations: Animal Care Services **ACS**, Principal Investigator **PI**, subcutaneous **SC**, intravenous **IV**, intraperitoneal **IP**, intramuscular **IM**, per os **PO**, per rectum **PR**

Use the following table to determine the most appropriate site for blood collection based on the volume required.

Site	Submandibular	Saphenous	Submental	Tail Vein	Retro-orbital	Cardiac Puncture
Multiple sampling	Yes	Yes	Yes	Yes	Yes	No
Volume	Max.200µl	Max. 200µl	Max. 200µl	50µl	Max. 200µl	TBV
Gauge Needle	4-5.5 mm lancet	23-25g	4-5.5 mm lancet	23-25g/scalpel	Capillary tube	23-25g

The following are "good practice" guidelines recommended for blood collection volumes, sites and needle gauges. As a general principle, sample volumes and number of samples should be kept to a minimum. As a general guide, up to 7.5% of the total blood volume can be taken on a single occasion from a normal, healthy animal on an adequate plane of nutrition with minimal adverse effects; 10% once every two weeks and 15% once every four weeks. For repeat bleeds at shorter intervals, a maximum of 1.0% of an animal's total blood volume can be removed every 24 hours. The acceptable quantity and frequency of blood sampling is dependent on the circulating blood volume of the animal and the red blood cell (RBC) turnover rate (RBC life span of the mouse: 38-47 days / RBC life span of the rat: 42-65 days). Always taken into consideration must be:

- The species to be sampled
- The size of the animal to be sampled
- The age and health of the animal to be sampled
- The effects of handling stress
- The collection site
- The frequency of sampling necessary
- The training and experience of the personnel performing the collection
- The suitability of sedation and/or anesthesia
- The minimum volume required for analysis. *The maximum permitted blood volume includes blood lost during collection. As a general rule, 20 drops = 1 mL (i.e. 5 drops = 250 µL)*

When collecting blood it is very important that the handler is able to recognize signs of shock and anemia. The combined effect of sample volume and sample frequency without appropriate fluid replacement can cause an animal to go into hypovolaemic shock or become anemic. Packed cell volume, haemoglobin level, red blood cell and reticulocyte counts should be monitored

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throughout a series of bleeds using the results from the first sample from each animal as the baseline for the animal.

- Signs of hypovolemic shock include a fast and thready pulse, pale dry mucous membranes, cold skin and extremities, restlessness, hyperventilation, and a sub-normal body temperature.
- Signs of anemia include pale mucous membranes of the conjunctiva or inside the mouth, pale tongue, gums, ears or footpads (non-pigmented animals), intolerance to exercise and with severe anemia, increased respiratory rate when at rest.

If >10% blood volume is required, it is recommended to replace collected blood volume by 3–4 times the volume of blood collected with isotonic fluids (i.e. fluids with same tonicity as blood, such as 0.9% saline, 5% dextrose or Lactated Ringer's solution).

The Circulating Blood Volume (CBV) of an adult mouse is ~72 ml/kg (0.072ml/g).

- 1% (maximum) of the CBV can be collected every 24 hours.
- 7.5% (maximum) of the CBV can be collected in a single collection, once per week.
- 10% (maximum) of the CBV can be collected in a single collection, once per every 2 weeks.
- 15% (maximum) of the CBV can be collected in a single collection, once per every 4 weeks.

To calculate blood collection volumes:

Body weight x Circulating Blood Volume = Total Blood Volume (TBV)

- $TBV \times \% \text{ (based on desired frequency of collection)} = \text{allowable volume to be collected.}$
- **i.e. For a single collection once per week:** $20 \text{ g} \times 0.072 \text{ ml/g} = 1.44 \text{ ml/g}$ **then** $1.44 \times 0.075 \text{ (7.5\% for once per week sample)} = 0.1 \text{ ml}$ is the maximum allowable volume.

Body Weight (g)	Total Circulating Blood Volume (ml)	Acceptable volume for collection μl (ml)			
		1.0% cumulative or single collection every 24 hrs.	7.5% single collection once per week	10% single collection once per every 2 weeks	15% single collection once per every 4 weeks
15	1.08	11 μl	80 (0.08)	108 (0.11)	160 (0.16)
20	1.44	14 μl	108 (0.11)	144 (0.14)	216 (0.21)
25	1.80	18 μl	135 (0.14)	180 (0.18)	270 (0.27)
30	2.16	22 μl	162 (0.16)	216 (0.22)	300 (0.33)
35	2.52	25 μl	189 (0.19)	252 (0.25)	375 (0.37)
40	2.88	29 μl	216 (0.22)	288 (0.29)	430 (0.43)

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This schedule allows for recovery time for the animals as illustrated in the following table:

Percent of blood volume collected in a SINGLE sampling	Recovery period (weeks)	Percent of blood volume collected over a 24-HOUR PERIOD (MULTIPLE samples)	Recovery period (weeks)
7.5%	1	7.5%	1
10%	2	10 - 15%	2
15%	4	20%	4

2. Materials:

- Sterile syringes
- Sterile needles (multiple sizes ranging from 23-30g)
- Gauze
- Alcohol swabs
- Collection tubes
- Anaesthetics
- Surgical tools as needed

3. Procedures:

- Only University Animal Care Committee (UACC) approved blood collection techniques can be performed.
- The minimal volume required should be collected at all times.
- All collections should be performed by trained and competent individuals.
- The smallest needle size for the collection location (avoiding hemolysis) should be used.
- Each and every animal requires a new sterile syringe and a new sterile needle/lancet. Prepare all equipment in advance.
- Only three attempts per site should be practiced. If unsuccessful, allow another trained person to collect the sample.
- Apply pressure with gauze until hemostasis occurs.

Cardiac Puncture

- Each and every animal requires a new sterile syringe and a new sterile needle. This is a terminal procedure.
 - There are three methods. Dorsal recumbency (lateral and ventral aspirate) and lateral recumbency.
 - Anesthetize the mouse following the *SOP 7.6 "Anaesthesia in Mice"*.
 - When sterility is of concern, the skin surrounding the desired puncture site can be excised to expose the underlying muscle area for needle insertion.
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- Once the animal has reached a surgical plane of anaesthesia, lay the animal either in dorsal recumbency or lateral recumbency.
- Release the vacuum on the syringe prior to skin puncture.

Dorsal Recumbency (Lateral aspirate)

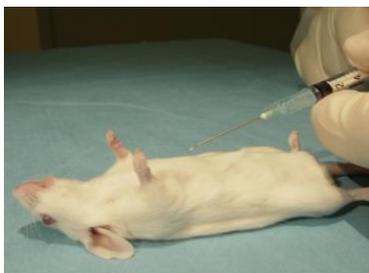
- Using the elbow to help indicate location along the rib cage, palpate for a strong heartbeat.
- Insert the needle bevel up into the thoracic cavity at 15-20angle directly lateral to the midline.
- When there is evidence of blood within the hub of the needle, steady the syringe and withdraw blood slowly.

Dorsal Recumbency (Ventral aspirate)

- From the sternum trace down the centre of the ribcage and locate the xyphoid process of the rib cage. Allow your needle to "fall" below this landmark.
- Insert the needle bevel up into the sternum and angle the syringe approximately 30 degrees cranially or towards the strongest heartbeat.
- When there is evidence of blood within the hub of the needle, steady the syringe and withdraw blood slowly.

Lateral Recumbency

- Place the animal on its right side facing down.
- Using the elbow to help indicate location along the rib cage, palpate for a strong heartbeat.
- Insert the needle into the thoracic cavity where the heart beat is the strongest.
- When there is evidence of blood within the hub of the needle, steady the syringe and withdraw blood slowly.



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<https://doi.org/10.1371/journal.pone.0113225>

SOP Revision History:

Date	New Version
9/22/2025	Triennial review
1/25/2018	Triennial review
2/28/2019	Updated SOP
2/28/2022	Triennial review
7/18/2022	Original SOP separated into different blood collections SOPs