



OFFICE OF ANIMAL WELFARE

Research Support Services

Date: February 18, 2016

To: Dr. David M. Anderson
Executive Director
Health Sciences Administration
Box 356355

Subject: Semiannual Program and Facility Review of Animal Use Program by the IACUC

The attached report represents the semiannual report of the Institutional Animal Care and Use Committee (IACUC) for the period of July 1, 2015 through December 31, 2015.

Please let me know if you have any questions about the content of this report.

Sincerely,

Tena Petersen
Manager, Regulatory Programs
Office of Animal Welfare

cc: Dr. Sally Thompson-Iritani
Director, Office of Animal Welfare

Dr. Jane Sullivan
Chair, Institutional Animal Care and Use Committee



UNIVERSITY OF WASHINGTON
OFFICE OF ANIMAL WELFARE

Date: January 21, 2016

To: University of Washington Institutional Animal Care and Use Committee

From: Tena Petersen
Manager of Regulatory Programs

Subject: Summary of items identified as part of the Semiannual Program and Facility Review of Animal Use Program by the IACUC

This represents the semiannual report of the Institutional Animal Care and Use Committee (IACUC) for the period of July 1, 2015 through December 31, 2015 as required by the PHS Policy on Humane Care and Use of Laboratory Animals and as a condition of this institution's Animal Welfare Assurance on file with the Office of Laboratory Animal Welfare (OLAW), and USDA Animal Welfare Regulations, 9 CFR Chapter I, subchapter A, as applicable. There were no minority opinions filed during this reporting period.

Evaluation of the Animal Care and Use Program and Inspection of Facilities

The IACUC has conducted its semiannual evaluation of the Institution's Animal Care and Use Program, using the 2011 8th edition of the Guide for the Care and Use of Laboratory Animals (Guide), and, as applicable, 9 CFR Chapter I, 2.31.

A brief overview of the all deficiencies identified during this reporting period is provided below and more detailed information is included in the attached summary (**Appendix A**). A deficiencies analysis is also attached (**Appendix B**). Corrections for deficiencies noted during the current reporting period have been completed or are in progress.

Significant Deficiencies

There was 1 Significant Deficiency identified during this reporting period. This deficiency was found in the category of Protocol and was due to the failure to administer analgesics according to the IACUC protocol. This is a decrease of 2 deficiencies from the last reporting cycle.

Minor Deficiencies

There were 476 deficiencies that were individually classified as "minor".

Events Reported to the IACUC during this reporting period.

- ❖ There were 7 adverse events that were reported to the IACUC and Institutional Official. All of these items were also reported to OLAW.

- ❖ There were 11 Facility, Temperature and Humidity events and 1 power related event reported to the IACUC.

Animal Welfare Concern:

There were no animal welfare concerns reported to the IACUC during this reporting period.

Site Visit Deficiency Summary

1. Site Visit Deficiency Summary
See **Appendix A**
2. Analysis of Deficiencies Found
See **Appendix B**

IACUC Protocols active during the semi-annual period with multiple major surgery, potential unrelieved pain or exceptions to the Guide or USDA Animal Welfare Regulations:

Multiple Major Surgeries

Performance of multiple major operative procedures – There were 60 protocols approved for multiple major operative procedures.

See **Appendix C**

Harm Benefit Analysis Subcommittee Report

During the past cycle the HBAS reviewed protocols in order to evaluate the harm benefit of the proposal and inform the process for categorization of the protocol. Protocols that have been discussed include Food and Water Restriction, Bone Marrow Transplant, Forced Swim Test, Foot Shock. Additionally, the Committee reviews clinical and research related unexpected events in order to inform the IACUC as well as assessing the harm benefit of each event. This process is on-going and will continue to review and evaluate protocols on a regular basis.

Exceptions to the Guide, USDA Animal Welfare Regulations and UW Policies

1. IACUC approved variances – There are five types of variances that have been approved on animal use protocols. There are approximately:
 - 18 Cage type variances
 - 11 temperature variances
 - 14 wire bottom cages
 - 41 weaning variances
 - 5 Policy variancesSee **Appendix D**
2. Environmental Enrichment Exemptions – There are 156 EEC exemptions that were reviewed and approved by the IACUC.
3. IACUC approved cage size variances for USDA species – There is one cage size variance for non-human primates. The IACUC approved an exemption to house infant monkeys (age 2 weeks to a body weight of 1 kg) in cages that meet floor space requirements but are only 24" high. These animals climb up well, but are not adept at climbing down. The lower cage height reduces the risk of injury if an animal should fall.

4. IACUC approved policy with exceptions to the Guide – Sanitation Schedule for Rodent Cage Wire Tops and Filter Tops
5. Exemptions approved by IACUC and USDA – There was one protocol (2340-01) that was approved for a USDA exemption to allow for up to 5 major operative procedures involving cranial implants for animal A09044. This exemption is no longer approved by the IACUC.

Policies/SOPs Reviewed by the IACUC

1. Analgesia In Research Animals, August 20, 2015, December 17, 2015
2. Anesthesia Certification Requirements in USDA Species, August 20, 2015, November 19, 2015
3. B.1068 Environmental Enrichment for Cats, October 22, 2015
4. B.1069 Environmental Enrichment for Ferrets, October 22, 2015
5. B.1070 Environmental Enrichment for Swine, October 22, 2015
6. B.1071 Environmental Enrichment for Rats, October 22, 2015
7. B.1072 Environmental Enrichment for Mice, October 22, 2015
8. B.1084 Environmental Enrichment for Aquatic Amphibians (*Xenopus laevis*), October 22, 2015,
9. B.1086 Environmental Enrichment for Guinea Pigs, October 22, 2015
10. B.1095 Environmental Enrichment for Gerbils, October 22, 2015,
11. B.1096 Environmental Enrichment for Chicks and Poults, October 22, 2015
12. B.1097 Environmental Enrichment for Bats, October 22, 2015
13. B.1098 Environmental Enrichment for Hamsters, October 22, 2015
14. B.1099 Environmental Enrichment for Birds, October 22, 2015
15. Cage Sanitization Policy, July 23, 2015
16. Food and Water Restriction in Mice and Rats, December 17, 2015
17. NHP Social Contact Introduction Procedure, November 19, 2015
18. Non-Pharmaceutical Grade Substances in Laboratory Animals, December 17, 2015
19. Number of Mice Maintained in a Breeding Cage, August 20, 2015
20. Recommended Analgesics, August 20, 2015
21. Rodent Cage Sanitation Frequency Policy, August 20, 2015
22. Veterinary Verification and Consultation of Timing of Procedures, Timing of Administration of Agents, and Changes to Procedures, October 22, 2015, December 17, 2015

IACUC Member Training

1. Harm Benefit Analysis, July 23, 2015
2. Review of Adverse Events on IACUC Protocols, August 20, 2015
3. Following Up on Concerns During IACUC Site Visits, August 20, 2015
4. AAALAC Site Visit Process and Common Deficiencies, October 22, 2015
5. Preparation, Handling and Use of MS222, December 17, 2015

Semiannual Program Review Checklist

Institutional Policies and Responsibilities

Semi-Annual Period: July 1, 2015 – December 31, 2015

1. Animal Care and Use Program	A*	M	S	C	NA
• Responsibility for animal well-being is assumed by all members of the program (<i>Guide</i> , p 1) [must]	X				
• IO has authority to allocate needed resources (<i>Guide</i> , p 13)	X				
• Resources necessary to manage program of veterinary care are provided (<i>Guide</i> , p 14) [must]	X				
• Sufficient resources are available to manage the program, including training of personnel in accord with regulations and the <i>Guide</i> (<i>Guide</i> , pp 11, 15)	X				
• Program needs are regularly communicated to IO by AV and/or IACUC (<i>Guide</i> , p 13)	X				
• Responsibilities for daily animal care and facility management are assigned to specific individual(s) when a full-time veterinarian is not available on site (<i>Guide</i> , p 14) [must]					X
• Inter-institutional collaborations are described in formal written agreements (<i>Guide</i> , p 15)	X				
• Written agreements address responsibilities, animal ownership, and IACUC oversight (<i>Guide</i> , p 15)	X				
2. Disaster Planning and Emergency Preparedness	A*	M	S	C	NA
• Disaster plans for each facility to include satellite locations are in place (<i>Guide</i> , p 35, p 75) [must]	X				
• Plans include provisions for euthanasia (<i>Guide</i> , p 35) [must]	X				
• Plans include triage plans to meet institutional and investigators' needs (<i>Guide</i> , p 35)	X				
• Plans define actions to prevent animal injury or death due to HVAC or other failures (<i>Guide</i> , p 35)	X				
• Plans describe preservation of critical or irreplaceable animals (<i>Guide</i> , p 35)	X				
• Plans include essential personnel and their training (<i>Guide</i> , p 35)	X				
• Animal facility disaster plans are approved by the institution and incorporated into the overall institutional response plan (<i>Guide</i> , p 35)	X				
• Law enforcement and emergency personnel are provided a copy and integration with overall plan is in place (<i>Guide</i> , p 35)	X				
3. IACUC	A*	M	S	C	NA
• Meets as necessary to fulfill responsibilities (<i>Guide</i> , p 25) [must]	X				
• IACUC Members named in protocols or with conflicts recuse themselves from protocol decisions (<i>Guide</i> , p 26) [must]	X				
• Continuing IACUC oversight after initial protocol approval is in place (<i>Guide</i> , p 33)	X				
• IACUC evaluates the effectiveness of training programs (<i>Guide</i> , p 15)	X				

4. IACUC Protocol Review - Special Considerations	A*	M	S	C	NA
• Humane endpoints are established for studies that involve tumor models, infectious diseases, vaccine challenge, pain modeling, trauma, production of monoclonal antibodies, assessment of toxicologic effects, organ or system failure, and models of cardiovascular shock (<i>Guide</i> , p 27)	X				
• For pilot studies, a system to communicate with the IACUC is in place (<i>Guide</i> , p 28)	X				
• For genetically modified animals, enhanced monitoring and reporting is in place (<i>Guide</i> , p 28)	X				
• Restraint devices are justified in the animal use protocols (<i>Guide</i> , p 29) [must]	X				
• Alternatives to physical restraint are considered (<i>Guide</i> , p 29)	X				
• Period of restraint is the minimum to meet scientific objectives (<i>Guide</i> , p 29)	X				
• Training of animals to adapt to restraint is provided (<i>Guide</i> , p 29)	X				
• Animals that fail to adapt are removed from study (<i>Guide</i> , p 29)	X				
• Appropriate observation intervals of restrained animals are provided (<i>Guide</i> , p 29)	X				
• Veterinary care is provided if lesions or illness result from restraint (<i>Guide</i> , p 30) [must]	X				
• Explanations of purpose and duration of restraint are provided to study personnel (<i>Guide</i> , p 30)	X				
• Multiple surgical procedures on a single animal are justified and outcomes evaluated (<i>Guide</i> , p 30)	X				
• Major versus minor surgical procedures are evaluated on a case-by-case basis (<i>Guide</i> , p 30)	X				
• Multiple survival procedure justifications in non-regulated species conform to regulated species standards (<i>Guide</i> , p 30)	X				
• Animals on food/fluid restriction are monitored to ensure nutritional needs are met (<i>Guide</i> , p 31)	X				
• Body weights for food/fluid restricted animals are recorded at least weekly (<i>Guide</i> , p 31)	X				
• Daily written records are maintained for food/fluid restricted animals (<i>Guide</i> , p 31)	X				
• Pharmaceutical grade chemicals are used, when available, for animal-related procedures (<i>Guide</i> , p 31) unless an exception is approved by the IACUC (see next item)	X				
• Non-pharmaceutical grade chemicals are described, justified, and approved by IACUC (<i>Guide</i> , p 31)	X				
• Investigators conducting field studies know zoonotic diseases, safety issues, laws and regulations applicable in study area (<i>Guide</i> , p 32)	X				
• Disposition plans are considered for species removed from the wild (<i>Guide</i> , p 32)	X				
• Toe-clipping is only used when there is no alternative and is performed aseptically and with pain relief (<i>Guide</i> , p 75)	X				

5. IACUC Membership and Functions	A*	M	S	C	NA
• The IACUC is comprised of at least 5 members, appointed by CEO (PHS Policy, IV.A.3.)	X				
• Members include a veterinarian, a scientist, a nonscientist, and a nonaffiliated non-lab animal user (<i>Guide</i> , p 24)	X				
• The IACUC has authority and is provided the resources for oversight and evaluation of the institution's program (<i>Guide</i> , p 14)	X				
• The IACUC conducts semiannual evaluations of the institutional animal care and use program (PHS Policy, IV.B.)	X				
• The IACUC conducts semiannual inspections of institutional animal facilities (PHS Policy, IV.B.)	X				
• The IACUC organizationally reports to the Institutional Official (PHS Policy, IV.A.1.b.)	X				
• Methods for reporting and investigating animal welfare concerns are in place (<i>Guide</i> , p 23) [must]	X				
• The IACUC reviews and investigates concerns about animal care and use at the institution (PHS Policy, IV.B.)	X				
• Procedures are in place for review, approval, and suspension of animal activities (PHS Policy, IV.B.)	X				
• Procedures are in place for review and approval of significant changes to approved activities (PHS Policy, IV.B.)	X				
• Policies are in place for special procedures (e.g., genetically modified animals, restraint, multiple survival surgery, food and fluid regulation, field investigations, agricultural animals) (<i>Guide</i> , p 27-32)	X				
• Requests for exemptions from the USDA's major survival surgical restrictions are made to USDA/APHIS (<i>Guide</i> , p 32) [must]	X				
6. IACUC Training	A*	M	S	C	NA
• All IACUC members receive:					
○ Formal orientation to institution's program (<i>Guide</i> , p 17)	X				
○ Training on legislation, regulations, guidelines, and policies (<i>Guide</i> , p 17)	X				
○ Training on how to inspect facilities and labs where animal use or housing occurs (<i>Guide</i> , p 17)	X				
○ Training on how to review protocols as well as evaluate the program (<i>Guide</i> , p 17)	X				
○ Ongoing training/education (<i>Guide</i> , p 17)	X				

7. IACUC Records and Reporting Requirements

A* M S C NA

• Semiannual report to the IO (PHS Policy, <u>IV.B.</u>)					
○ Submitted to IO every 6 months	X				
○ Compiles program review and facility inspection(s) results (includes all program and facility deficiencies)	X				
○ Includes minority IACUC views	X				
○ Describes IACUC-approved departures from the <i>Guide</i> or PHS Policy and the reasons for each departure	X				
○ Distinguishes significant from minor deficiencies	X				
○ Includes a plan and schedule for correction for each deficiency identified	X				
• Reports to OLAW (PHS Policy, <u>IV.F.</u>)					
○ Annual reports to OLAW document program changes, dates of the semiannual program reviews and facility inspections, and includes any minority views	X				
○ The institution promptly advises OLAW of serious/ongoing <i>Guide</i> deviations or PHS Policy noncompliance (NOT-OD-05-034)	X				
○ The Institution promptly advises OLAW of any suspension of an animal activity by the IACUC (NOT-OD-05-034)	X				
• Reports to U.S. Department of Agriculture (USDA) or Federal funding agency					
○ Annual reports to the USDA contain required information including all required reports of exceptions/exemptions and/or exceptions to the regulations and standards	X				
○ Reports are filed with the USDA within 15 days for failures to adhere to the timetable for correction of significant deficiencies involving USDA covered species	X				
○ Suspensions of activities by the IACUC are promptly reported to the USDA and any Federal funding agency	X				
○ Reporting mechanism to USDA is in place for IACUC-approved exceptions to the regulations and standards	X				
• Records (PHS Policy, <u>IV.E.</u>)					
○ IACUC meeting minutes and semiannual reports to the IO are maintained for 3 years	X				
○ Records of IACUC reviews of animal activities include all required information	X				
○ Records of IACUC reviews are maintained for 3 years after the completion of the study	X				

8. Veterinary Care (See also next section - Veterinary Care)	A*	M	S	C	NA
• An arrangement for veterinarian(s) with training or experience in lab animal medicine is in place including backup veterinary care	X				
• Veterinary access to all animals is provided (<i>Guide</i> , p 14) [must]	X				
• Direct or delegated authority is given to the Attending Veterinarian to oversee all aspects of animal care and use (<i>Guide</i> , p 14) [must]	X				
• A qualified veterinarian provides consultation when pain and distress exceeds the anticipated level in the protocol (<i>Guide</i> , p 5) [must]	X				
• Veterinarian provides consultation when interventional control is not possible (<i>Guide</i> , p 5) [must]	X				
• If there is only a part-time/consulting veterinarian, visits meet programmatic needs (<i>Guide</i> , p 14)					X
• Regular communication occurs between veterinarian and IACUC (<i>Guide</i> , p 14)	X				
• Veterinarian(s) have experience and training in species used (<i>Guide</i> , p 15) [must]	X				
• Veterinarian(s) have experience in facility administration/management (<i>Guide</i> , p 15)	X				
9. Personnel Qualifications and Training	A*	M	S	C	NA
• All personnel are adequately educated, trained, and/or qualified in basic principles of laboratory animal science. Personnel included: [must]					
○ Veterinary/other professional staff (<i>Guide</i> , p 15-16)	X				
○ IACUC members (<i>Guide</i> , p 17)	X				
○ Animal care personnel (<i>Guide</i> , p 16)	X				
○ Research investigators, instructors, technicians, trainees, and students (<i>Guide</i> , pp 16-17)	X				
• Continuing education for program and research staff provided to ensure high quality care and reinforce training (<i>Guide</i> , pp 16-17)	X				
• Training is available prior to starting an animal activity (<i>Guide</i> , p 17)	X				
• Training is documented (<i>Guide</i> , p 15)	X				
• Training program content includes: (<i>Guide</i> , p 17)					
○ Methods for reporting concerns (<i>Guide</i> , p 17)	X				
○ Humane practices of animal care (e.g., housing, husbandry, handling)	X				
○ Humane practices of animal use (e.g., research procedures, use of anesthesia, pre- and post-operative care, aseptic surgical techniques and euthanasia (<i>Guide</i> , p 17)	X				
○ Research/testing methods that minimize numbers necessary to obtain valid results (PHS Policy, IV.A.1.g.)	X				
○ Research/testing methods that minimize animal pain or distress (PHS Policy, IV.A.1.g.)	X				
○ Use of hazardous agents, including access to OSHA chemical hazard notices where applicable (<i>Guide</i> , p 20)	X				
○ Animal care and use legislation (<i>Guide</i> , p 17)	X				
○ IACUC functions (<i>Guide</i> , p 17)	X				
○ Ethics of animal use and the Three R's (<i>Guide</i> , p 17)	X				

10. Occupational Health and Safety of Personnel

A* M S C NA

	A*	M	S	C	NA
• Program is in place and is consistent with federal, state, and local regulations (<i>Guide</i> , p 17) [must]	X				
• Program covers <i>all</i> personnel who work in laboratory animal facilities (<i>Guide</i> , p 18)	X				
• Changing, washing, and showering facilities are available as appropriate (<i>Guide</i> , p 19)	X				
• Facilities where hazardous agents are used are separated from other areas and identified as limited access (<i>Guide</i> , p 19)	X				
• Personnel training (e.g., on zoonoses, hazards, personal hygiene, special precautions, animal allergies) is provided based on risk (<i>Guide</i> , p 20)	X				
• Personal hygiene procedures are in place (e.g., work clothing, eating/drinking/smoking policies) (<i>Guide</i> , p 20)	X				
• Procedures for use, storage, and disposal of hazardous biologic, chemical, and physical agents are in place (<i>Guide</i> , p 21)	X				
• Personal Protective Equipment for the work area is appropriate and available (<i>Guide</i> , p 21)	X				
• The program for medical evaluation and preventive medicine for personnel includes:					
○ Pre-employment evaluation including health history (<i>Guide</i> , p 22)	X				
○ Immunizations as appropriate (e.g., rabies, tetanus) and tests as appropriate (<i>Guide</i> , p 22)	X				
○ Zoonosis surveillance as appropriate (e.g., Q-fever, tularemia, Hantavirus, plague) (<i>Guide</i> , p 23)	X				
○ Procedures for reporting and treating injuries, including accidents, bites, allergies, etc. (<i>Guide</i> , p 23)	X				
○ Promotes early diagnosis of allergies including preexisting conditions (<i>Guide</i> , p 22)	X				
○ Considers confidentiality and other legal factors as required by federal, state and local regulations (<i>Guide</i> , p 22) [must]	X				
○ If serum samples are collected, the purpose is consistent with federal and state laws (<i>Guide</i> , p 22) [must]	X				
• Waste anesthetic gases are scavenged (<i>Guide</i> , p 21)	X				
• Hearing protection is provided in high noise areas (<i>Guide</i> , p 22)	X				
• Respiratory protection is available when performing airborne particulate work (<i>Guide</i> , p 22)	X				
• Special precautions for personnel who work with nonhuman primates, their tissues or body fluids include:					
○ Tuberculosis screening provided for all exposed personnel (<i>Guide</i> , p 23)	X				
○ Training and implementation of procedures for bites, scratches, or injuries associated with macaques (<i>Guide</i> , p 23)	X				
○ PPE is provided including gloves, arm protection, face masks, face shields, or goggles (<i>Guide</i> , p 21)	X				
○ Injuries associated with macaques are carefully evaluated and treatment implemented (<i>Guide</i> , p 23)	X				
• Occupational safety and health of field studies is reviewed by the Department of Environmental Health and Safety's "Research and Occupational Safety Section" (<i>Guide</i> , p 32)	X				

11. Personnel Security	A*	M	S	C	NA
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<ul style="list-style-type: none"> Preventive measures in place include pre-employment screening, and physical and IT security (<i>Guide</i>, p 23) 	X				
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12. Investigating & Reporting Animal Welfare Concerns	A*	M	S	C	NA
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<ul style="list-style-type: none"> Methods for investigating and reporting animal welfare concerns are established (<i>Guide</i>, p 23) [must] 	X				
<ul style="list-style-type: none"> Reported concerns and corrective actions are documented (<i>Guide</i>, p 24) 	X				
<ul style="list-style-type: none"> Mechanisms for reporting concerns are posted in facilities and on applicable websites with instructions (<i>Guide</i>, p 24) <ul style="list-style-type: none"> Includes multiple contacts (<i>Guide</i>, p 24) Includes anonymity, whistle blower policy, nondiscrimination and reprisal protection (<i>Guide</i>, p 24) 	X				

- * **A** = acceptable; **M** = minor deficiency; **S** = significant deficiency (is or may be a threat to animal health or safety)
C = change in program (PHS Policy IV.A.1.a.-i.) (include in semiannual report to IO and in annual report to OLAW)
NA = not applicable

NOTES:

Veterinary Care

Semi-Annual Period: July 1, 2014 – December 31, 2014

1. Clinical Care and Management	A*	M	S	C	NA
• The veterinary care program offers high quality of care and ethical standards (<i>Guide</i> , p 105) [must]	X				
• The Attending Veterinarian, through the Veterinary Care Program and Training Programs, provides guidance to all personnel to ensure appropriate husbandry, handling, treatment, anesthesia, analgesia, and euthanasia (<i>Guide</i> , p 106)	X				
• The Attending Veterinarian, through the Veterinary Care Program and Training Programs, provides oversight to surgery and perioperative care (<i>Guide</i> , p 106)	X				
• Veterinary care program is appropriate for program requirements (<i>Guide</i> , pp 113-114)	X				
• Veterinarian(s) is familiar with species and use of animals and has access to medical and experimental treatment records (<i>Guide</i> , p 114)	X				
• Procedures to triage and prioritize incident reports are in place (<i>Guide</i> , p 114)	X				
• Procedures are in place to address:					
○ Problems with experiments to determine course of treatment in consultation with investigator(<i>Guide</i> , p 114)	X				
○ Recurrent or significant health problems with the IACUC, and for documentation of treatments and outcomes (<i>Guide</i> , p 114)	X				
○ Veterinary review and oversight of medical and animal use records (<i>Guide</i> , p 115)	X				
• Procedures are established for timely reporting of animal injury, illness, or disease (<i>Guide</i> , p 114) [must]	X				
• Procedures are established for veterinary assessment, treatment, or euthanasia (<i>Guide</i> , p 114) [must]	X				
• Veterinarian is authorized to treat, relieve pain, and/or euthanize (<i>Guide</i> , p 114) [must]	X				

2. Animal Procurement and Transportation/Preventive Medicine	A*	M	S	C	NA
• Procedures for lawful animal procurement are in place (<i>Guide</i> , p 106) [must]	X				
• Sufficient facilities and expertise are confirmed prior to procurement (<i>Guide</i> , p 106)	X				
• Procurement is linked to IACUC review and approval (<i>Guide</i> , p 106)	X				
• Random source dogs and cats are inspected for identification (<i>Guide</i> , p 106)					X
• Population status of wildlife species is considered prior to procurement (<i>Guide</i> , p 106)	X				
• Appropriate records are maintained on animal acquisition (<i>Guide</i> , p 106)	X				
• Animal vendors are evaluated to meet program needs and quality (<i>Guide</i> , p 106)	X				
• Breeding colonies are based on need and managed to minimize numbers (<i>Guide</i> , p 107)	X				
• Procedures for compliance with animal transportation regulations, including international requirements, are in place (<i>Guide</i> , p 107) [must]	X				
• Transportation is planned to ensure safety, security and minimize risk (<i>Guide</i> , p 107)	X				
• Movement of animals is planned to minimize transit time and deliveries are planned to ensure receiving personnel are available (<i>Guide</i> , pp 107- 108)	X				
• Appropriate loading and unloading facilities are available (<i>Guide</i> , p 109)	X				
• Environment at receiving site is appropriate (<i>Guide</i> , p 109)	X				
• Policies are in place on separation by species, source, and health status (<i>Guide</i> , pp 109, 111-112)	X				
• Procedures are in place for quarantine and include zoonoses prevention (<i>Guide</i> , p 110)	X				
• Quarantined animals from different shipments are handled separately or physically separated (<i>Guide</i> , p 110)	X				
• Procedures are in place for stabilization/acclimation (<i>Guide</i> , pp 110-111)	X				
• Policies are in place for isolation of sick animals (<i>Guide</i> , p 112)	X				
• Program is in place for surveillance, diagnosis, treatment and control of disease, and includes daily observation (<i>Guide</i> , p 112)	X				
• Diagnostic resources are available for a preventive health program (<i>Guide</i> , p 112)	X				

3. Surgery	A*	M	S	C	NA
• Surgical outcomes are assessed to ensure that appropriate procedures are followed and that timely corrective changes are instituted (<i>Guide</i> , p 115)	X				
• Researchers have appropriate training to ensure good technique (<i>Guide</i> , p 115) [must]	X				
• Pre-surgical plans are developed and include veterinary input (e.g., location, supplies, anesthetic and analgesic use, peri-operative care, recordkeeping) (<i>Guide</i> , p 116)	X				
• Aseptic surgery is conducted in dedicated facilities or spaces, unless exception justified and IACUC approved (<i>Guide</i> , p 116)	X				
• Surgical procedures, including laparoscopic procedures, are categorized as major or minor (<i>Guide</i> , pp 117-118)	X				
• For nonsurvival surgery, the surgical site is clipped, gloves are worn and instruments and the area are clean (<i>Guide</i> , p 118)	X				
• Aseptic technique is followed for survival surgical procedures (<i>Guide</i> , pp 118-119)	X				
• Effective procedures for sterilizing instruments and monitoring expiration dates on sterile packs are in place (<i>Guide</i> , p 119)	X				
• Procedures for monitoring surgical anesthesia and analgesia are in place (<i>Guide</i> , p 119)	X				
• For aquatic species, skin surfaces are kept moist during surgical procedures (<i>Guide</i> , p 119)	X				
• Post-operative monitoring and care are provided by trained personnel and documented (e.g., thermoregulation, physiologic function, analgesia, infection, removal of skin closures) (<i>Guide</i> , pp 119-120)	X				
4. Pain, Distress, Anesthesia and Analgesia	A*	M	S	C	NA
• Guidelines for assessment and categorization of pain, distress and animal wellbeing are provided during training (<i>Guide</i> , p 121)	X				
• Selection of analgesics and anesthetics is based on professional veterinary judgment (<i>Guide</i> , p 121)	X				
• Painful procedures are monitored to ensure appropriate analgesic management (<i>Guide</i> , p 122)	X				
• Non-pharmacologic control of pain is considered as an element of post-procedural care (<i>Guide</i> , p 122)	X				
• Procedures are in place to assure antinoception before surgery begins (<i>Guide</i> , p 122) [must]	X				
• Guidelines for selection and use of analgesics and anesthetics are in place and are regularly reviewed and updated (<i>Guide</i> , p 122)	X				
• Special precautions for the use of paralytics are in place to ensure anesthesia (<i>Guide</i> , p 123)	X				

5. Euthanasia	A*	M	S	C	NA
• Methods are consistent with AVMA Guidelines on Euthanasia unless approved by the IACUC (<i>Guide</i> , p 123)	X				
• Standardized methods that avoid distress and consider animal age and species are developed and approved by the Attending Veterinarian and IACUC (<i>Guide</i> , pp 123-124)	X				
• Training is provided on appropriate methods for each species, and psychological stress to personnel performing the procedures is considered (<i>Guide</i> , p 124)	X				
• Procedures and training are in place to ensure death is confirmed (<i>Guide</i> , p 124) [must]	X				
6. Drug Storage and Control	A*	M	S	C	NA
• Program complies with federal regulations for human and veterinary drugs (<i>Guide</i> , p 115) [must]	X				
• Drug records and storage procedures are reviewed during semi-annual IACUC facility inspections (<i>Guide</i> , p 115)	X				
• Procedures are in place to ensure that analgesics and anesthetics are used within their expiration dates (<i>Guide</i> , p 122) [must]	X				
• Anesthetics and analgesics are acquired, stored, and their use and disposal are recorded legally and safely (<i>Guide</i> , p 122)	X				

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NA = not applicable

NOTES:

Semiannual Facility Inspection Checklist

Terrestrial Animal Housing and Support Areas

Date: July 1, 2015 – December 31, 2015

Location: Multiple

	A*	M	S	C	NA
• Location:					
○ animal areas separate from personnel areas (<i>Guide, p 134</i>)	X				
○ separation of species (<i>Guide, p 111</i>)	X				
○ separation by disease status (<i>Guide, p 111</i>)	X				
○ security and access control (<i>Guide, p 151</i>)	X				
• Construction:					
○ corridors (<i>Guide, p 136</i>)	X				
○ animal room doors (<i>Guide, p 137</i>) – some doors required minor repairs. One door required the instillation of a door sweep.		X			
○ exterior windows (<i>Guide, p 137</i>)	X				
○ floors (<i>Guide, p 137</i>) – Various floors require repairs.		X			
○ drainage (<i>Guide, p 138</i>)	X				
○ walls and ceilings (<i>Guide, p 138</i>) – Various walls and ceilings require repairs.		X			
○ heating ventilation and air conditioning (<i>Guide, p 139</i>)	X				
○ power and lighting (<i>Guide, p 141</i>)	X				
○ noise control (<i>Guide, p 142</i>)	X				
○ vibration control (<i>Guide, p 142</i>)	X				
○ environmental monitoring (<i>Guide, p 143</i>)	X				
• Room/Cage:					
○ temperature and humidity (<i>Guide, p 43</i>) – There were five deficiencies found related to temperature. The Attending Veterinarian reported several humidity issues to the IACUC as part of her report during IACUC meetings.		X			
○ ventilation and air quality (<i>Guide, p 45</i>)	X				
○ illumination (<i>Guide, p 47</i>) – In one room the back corner was found to be dark. A LED light system was ordered. In another room several lighted bulbs were not working and were replaced.		X			
○ noise and vibration (<i>Guide, p 49</i>) DCM SLU 3.1, E757, Interstitial blower fan noise is still too noisy (not the hood).	X				
• Primary Enclosure:					
○ space meets physiologic, behavioral i, and socialii needs (<i>Guide, pp 51, 55-63</i>)	X				
○ secure environment provided (<i>Guide, p 51</i>)	X				
○ durable, nontoxic materials in good repair and no risk of injury (<i>Guide, p 51</i>)	X				
○ flooring is safe and appropriate for species (<i>Guide, p 51</i>)	X				
○ adequate bedding and structures for resting, sleeping, breeding (<i>Guide, p 52</i>)	X				
○ objective assessments of housing and management are made (<i>Guide, p 52</i>)	X				
○ procedures for routine husbandry are documented (<i>Guide, p 52</i>) – in some areas husbandry logs were not properly completed.		X			
○ socially housed animals can escape or hide to avoid aggression (<i>Guide, p 55</i>)	X				
○ cage height provides adequate clearance (<i>Guide, p 56</i>)	X				
○ animals express natural postures, can turn around, access food and water, and rest away from urine and feces (<i>Guide, p 56</i>) [must]	X				
○ rationaleiii for <i>Guide</i> /USDA space exceptions approved by IACUC and based on performance indices (<i>Guide, p 56</i>)	X				
○ dogs and cats allowed to exercise and provided human interaction (<i>Guide, p 58</i>)	X				
○ nonhuman primates are socially housed except for scientific, veterinary or	X				

behavior reasons (<i>Guide, pp 58-59</i>)					
o single housing of nonhuman primates is for shortest duration possible (<i>Guide, p 60</i>)	X				
o opportunities for release into larger enclosures is considered for single caged nonhuman primates (<i>Guide, p 60</i>)	X				
o agricultural animals are housed socially (<i>Guide, p 60</i>)	X				
o food troughs and water devices for agricultural animals allow access for all animals (<i>Guide, p 60</i>)	X				
• Environmental Enrichment, Behavioral and Social Management:					
o structures and resources promote species typical behavior (<i>Guide, pp 52-54</i>)	X				
o novelty of enrichment is considered (<i>Guide, p 53</i>)	X				
o species specific plans for housing including enrichment, behavior and activity are developed and reviewed regularly by IACUC, researchers and veterinarian (<i>Guide, pp 53, 58, 60, 63</i>)	X				
o animal care personnel receive training to identify abnormal animal behaviors (<i>Guide, p 53</i>)	X				
o stability of pairs or groups is monitored for incompatibility (<i>Guide, p 64</i>)	X				
o single housing is justified for social species (<i>Guide, p 64</i>)	X				
o single housing is limited to the minimum period necessary (<i>Guide, p 64</i>)	X				
o additional enrichment for single housed animals is provided (<i>Guide, p 64</i>)	X				
o single housing is reviewed regularly by IACUC and veterinarian (<i>Guide, p 64</i>)	X				
o habituation to routine procedures is part of enrichment program (<i>Guide, p 64</i>)	X				
• Sheltered or Outdoor Housing: (e.g., barns, corrals, pastures, islands)					
o weather protection and opportunity for retreat (<i>Guide, p 54</i>) [must]	X				
o appropriate size (<i>Guide, p 54</i>)	X				
o ventilation and sanitation of shelter (no waste/moisture build-up) (<i>Guide, p 54</i>)	X				
o animal acclimation (<i>Guide, p 55</i>)	X				
o social compatibility (<i>Guide, p 55</i>)	X				
o roundup/restraint procedures (<i>Guide, p 55</i>)	X				
o appropriate security (<i>Guide, p 55</i>)	X				
• Naturalistic Environments:					
o animals added /removed with consideration of effect on group (<i>Guide, p 55</i>)					X
o adequate food, fresh water, and shelter ensured (<i>Guide, p 55</i>)					X
• Food:					
o feeding schedule and procedures including caloric intake management (<i>Guide, pp 65-67</i>) – In one case there was no indication as to when mice on food restriction were last fed. No food log present. DCM needs to check group's feeding log and document this change.		X			
o contamination prevention (<i>Guide, p 65</i>)	X				
o vendor quality control (<i>Guide, p 66</i>)	X				
o storage in sealed containers (<i>Guide, p 66</i>)	X				
o expiration date labeling (<i>Guide, p 66</i>) – Expired food or issues related to documentation of expiration dates were noted.		X			
o vermin control (<i>Guide, p 66</i>) In some cases food was not being stored 6" from the wall or was not stored in appropriate containers.		X			
o rotation of stocks (<i>Guide, p 66</i>)	X				
• Water:					
o ad libitum unless justified (<i>Guide, pp 67-68</i>)	X				
o QC procedures (<i>Guide, pp 67-68</i>)	X				
• Bedding and Nesting Materials:					
o species appropriate (<i>Guide, pp 68-69</i>)	X				
o keeps animals dry (<i>Guide, pp 68-69</i>)	X				
o QC procedures (<i>Guide, pp 68-69</i>)	X				
o minimizes scientific variables (<i>Guide, pp 68-69</i>)	X				

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 NA = not applicable

Aquatic Animal Housing and Support Areas

Date: January 1, 2015 – June 30, 2015

Location: Multiple

	A*	M	S	C	NA
• Location:					
o animal areas separate from personnel areas (Guide, p 134)	X				
o separation of species (Guide, p 111)	X				
o separation by disease status (Guide, p 111)	X				
o security and access control (Guide, p 151)	X				
• Construction:					
o corridors (Guide, p 136)	X				
o animal room doors (Guide, pp 137, 150)	X				
o exterior windows (Guide, p 137)	X				
o floors (Guide, pp 137, 150)	X				
o drainage (Guide, pp 138, 150)	X				
o walls and ceilings (Guide, pp 138, 150)	X				
o heating ventilation and air conditioning (Guide, pp 139, 150-151)	X				
o power and lighting (Guide, pp 141, 150)	X				
o noise control (Guide, p 142)	X				
o vibration control (Guide, p 142)	X				
o environmental monitoring (Guide, p 143)	X				
o Other – Earthquake bracing of water towers	X				
• Water Quality:					
o standards for acceptable quality are established (Guide, p 78)	X				
o chlorine, chloramines, chemical, and reactive bioproducts are removed or neutralized prior to use in aquatic systems (Guide, pp78, 86) [must]	X				
• Life Support System:					
o water source is based on appropriate controls and research requirements (Guide, p 79)	X				
o biofilter is of sufficient size to process bioload (Guide, p 80) [must]	X				
• Temperature, Humidity and Ventilation/Illumination/Noise and Vibration:					
o temperature and humidity (Guide, pp 43, 80-81)	X				
o ventilation and air quality (Guide, pp 45, 81)	X				
o illumination (Guide, pp 47, 81)	X				
o noise and vibration (Guide, pp 49, 81)	X				
• Primary Enclosure:					
o allows for normal physiological and behavioral needs (Guide, p 82)	X				
o allows social interaction for social species (Guide, p 82)	X				
o provides a balanced, stable environment (Guide, p 82)	X				
o provides appropriate water quality and monitoring (Guide, p 82)	X				
o allows access to food and waste removal (Guide, p 82)	X				
o restricts escape and entrapment (Guide, p 82)	X				
o allows undisturbed observation (Guide, p 82)	X				
o constructed of nontoxic materials (Guide, p 82)	X				
o prevents electrical hazards (Guide, p 82)	X				

o space needs of species are evaluated by IACUC during program evaluations and facility inspections (Guide, p 83)	X				
• Environmental Enrichment, Social Housing, Behavioral and Social Management:					
o enrichment elicits appropriate behaviors and is safe (Guide, p 83)	X				
o semi-aquatic reptiles are provided terrestrial areas (Guide, p 83)	X				
o handling is kept to a minimum and appropriate techniques are in place at facility or protocol level (Guide, p 84)	X				
o nets are cleaned, disinfected and managed to avoid contamination of systems (Guide, p 84)	X				
• Food:					
o storage to prevent contamination, preserve nutrients and prevent pests (Guide, p 84) Roosevelt 1110, Secondary container of fish food was not labeled with contents.	X				
o delivery ensures access to all, minimizing aggression and nutrient loss (Guide, p 84)	X				
o storage times are based on manufacturer recommendations or accepted practice (Guide, p 84) – Roosevelt 110, Expired bottle of cod liver oil and fish food flakes was found in the refrigerator.	X				
o a nutritionally complete diet is provided (Guide, p 84)	X				
• Substrate:					
o amount, type and presentation of substrate is appropriate for the system and the species (Guide, p 85)	X				
• Sanitation, Cleaning and Disinfection					
o frequency of tank/cage cleaning and disinfection is determined by water quality, permits adequate viewing and health monitoring (Guide, p 86)	X				
o cleaning and disinfection of macroenvironment (Guide, p 86)	X				
• Waste Disposal:					
o procedures for collection (Guide, pp 73-74)	X				
o hazardous wastes are rendered safe before removal from facility (Guide, pp 73-74) [must]	X				
o animal carcasses (Guide, pp 73-74)	X				
• Pest Control:					
o regularly scheduled (Guide, p 74)	X				
o documented program including control of pests and insecticide use (Guide, p 74)	X				
• Emergency, Weekend, and Holiday Animal Care:					
o care provided by qualified personnel every day (Guide, pp 74, 87)	X				
o provision for accessible contact information (Guide, pp 74, 87)	X				
o emergency response plans in place to address major system failures (Guide, 87)	X				
o veterinary care available after hours, weekends, and holidays (Guide, pp 74, 114) [must]	X				
• Identification:					
o cage/tank cards contain required information (Guide, pp 75, 87)	X				
o genotype information included and standardized nomenclature used when applicable (Guide, pp 75, 87)	X				
• Recordkeeping:					
o water quality parameters and frequency of testing recorded (Guide, p 88)	X				
o records kept on feeding, nonexpired food supplies, live cultures (Guide, p 88)	X				
• Storage:					
o adequate space for equipment, supplies, food, substrate and refuse (Guide, p 141)	X				
o substrate protected from contamination (Guide, p 141)	X				
o food in vermin-free, temperature and humidity controlled area and protected from contamination (Guide, p 141)	X				
o refuse storage is separate (Guide, p 141)	X				
o carcass and animal tissue storage is separate, refrigerated below 7°C and	X				

cleanable (Guide, p 141)					
• Personnel:	X				
○ adequate space for locker rooms, administration and training (Guide, p 135)	X				

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NA = not applicable

Cagewash

Date: January 1, 2015 – June 30, 2015

Location: Multiple

	A*	M	S	C	NA
• Construction and Operation:					
○ dedicated central area for sanitizing cages and equipment is provided (Guide, p 143)	X				
○ cage-washing equipment meets need (Guide, p 143)	X				
○ doors, windows, floors, drainage, walls, ceilings (Guide, pp 136-138) – Various walls and ceilings require repairs.		X			
○ convenient to animal areas/waste disposal (Guide, p 143)	X				
○ ease of access (including door size) facilitates use (Guide, p 143)	X				
○ sufficient space for staging and maneuvering (Guide, p 143)	X				
○ safety precautions/clothing/equipment used for waste disposal/prewash/acid wash ((Guide, p 143))	X				
○ traffic flow clean to dirty with no contamination of clean equipment by dirty equipment and appropriate air pressurization (Guide, p 143)	X				
○ insulation and/or sound attenuation present as needed (Guide, p 143)	X				
○ utilities are appropriate (Guide, p 143)	X				
○ ventilation meets heat and humidity load (Guide, p 143)	X				
○ safety features (e.g., SOPs, warning signs, eyewash stations) are in use (Guide, p 143) – 1) DCM 6 th Floor Vivarium E607, Personnel was not wearing earplugs in the Clean Side Cage Wash. 2) DCM HR&T Vivarium SB048, Eyewash station is not present in the Dirty side Cage wash room.		X			
○ functioning safety devices to prevent entrapment in washer/sterilizers (Guide, p 143) – There were 3 deficiencies noted related to inappropriate safety signage.			X		
○ cage wash temperatures are monitored and records are available (Guide, p 73)	X				
○ appropriate clean cage storage (Guide, p 141)	X				

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NA = not applicable

Special Facilities: Aseptic Surgery

Date: January 1, 2015 – June 30, 2015

Location: Multiple

	A*	M	S	C	NA
• General Considerations:					
○ location minimizes traffic/contamination (<i>Guide, p 144</i>)	X				
○ functional components (surgical support, animal preparation, surgeon scrub, operating room, postoperative recovery) are designed and separated (physically or otherwise) (<i>Guide, p 144</i>)	X				
○ appropriate drug storage, control, expiration date monitoring (<i>Guide, pp 115, 122</i>)	X				
○ safe sharps disposal system (<i>Guide, p 74</i>)	X				
○ adequate records of anesthesia and perioperative care (<i>Guide, p 122</i>)	X				
○ aseptic procedures in use for all survival surgery (<i>Guide, pp 118-119</i>)	X				
• Operating Room:					
○ effective contamination control procedures (<i>Guide, p 144</i>)	X				
○ effective cleaning procedures/dedicated tools (<i>Guide, p 145</i>)	X				
○ interior surfaces smooth and impervious to moisture (<i>Guide, p 145</i>)	X				
○ HVAC system meets <i>Guide</i> requirements (<i>Guide, p 145</i>)	X				
○ lighting safe and appropriate (<i>Guide, p 145</i>)	X				
○ outlets safe and appropriate (<i>Guide, p 145</i>)	X				
○ scavenging of anesthetic gases implemented (<i>Guide, p 145</i>)	X				
• Surgical Support:					
○ facility for washing, sterilizing, storing instruments and supplies (<i>Guide, p 145</i>)	X				
○ autoclave monitoring procedures are implemented (<i>Guide, pp 119, 145</i>) – In one case the autoclave was used but there was no sterilization strip logged for that date.		X			
○ storage of autoclaved materials maintains sterility (<i>Guide, p 145</i>) – 20 endotracheal tubes in autoclave pouches were past sterilization date. A “Sterilized for decontamination – non sterile use only” sticker was placed over the date during the visit.		X			
○ cold sterilization procedures are appropriate (<i>Guide, p 119</i>)	X				
• Animal Preparation: contains large sink to facilitate cleaning of animal and operative site (<i>Guide, p 145</i>)	X				
• Surgeon Scrub: outside operating room, non-hand-operated sink (<i>Guide, p 145</i>)	X				
• Postoperative Recovery: allows adequate observation, easily cleaned, supports physiologic functions, minimizes risk of injury (<i>Guide, p 145</i>)	X				
• Dressing Area: place for personnel to change (<i>Guide, p 145</i>)	X				

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NA = not applicable

Special Facilities: Procedure Areas, Non-survival Surgeries, Laboratories, Rodent Surgeries, Imaging, Whole Body Irradiation, Hazardous Agent Containment, Behavioral Studies

Date: January 1, 2015 – June 30, 2015

Location: Multiple

	A*	M	S	C	NA
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<ul style="list-style-type: none"> • General Considerations: 					
o labs used to house animals only when scientifically required and limited to minimum period necessary (<i>Guide, p 134</i>)	X				
o drug storage, control, and expiration dates (<i>Guide, pp 115, 122</i>)	X				
o sharps disposal (<i>Guide, p 74</i>)	X				
o anesthetic monitoring (<i>Guide, p 120</i>)	X				
o scavenging of anesthetic gases (<i>Guide, p 21</i>)	X				
o safety features (e.g., SOPs, safety signs, eyewash stations, secure gas cylinders) are in place (<i>Guide, p 19</i>) - in some cases compressed gas tanks were not properly secured.	X				
o carcass disposal (<i>Guide, pp 73-74</i>)	X				
<ul style="list-style-type: none"> • Additional Concerns for Survival Surgery: (rodent and minor procedures only) 					
o rodent survival surgery clean and uncluttered, not used for anything else during surgery (<i>Guide, p 144</i>)	X				
o records of peri-operative care (<i>Guide, p 120</i>)	X				
o aseptic procedures (<i>Guide, pp 118-119</i>)	X				
o autoclave monitoring procedures (<i>Guide, pp 119, 145</i>)	X				
o storage of autoclaved materials (<i>Guide, p 145</i>)	X				
o cold sterilization procedures are appropriate (<i>Guide, p 119</i>)	X				
<ul style="list-style-type: none"> • Imaging/Whole Body Irradiation: NEW 					
o location of resource limits contamination risk (<i>Guide, p 147</i>)	X				
o appropriate transportation methods are in place (<i>Guide, p 147</i>)	X				
o gas anesthesia provision, scavenging and monitoring are appropriate (<i>Guide, p 147</i>)	X				
o appropriate sensors and ventilation are provided for cryogen gases (<i>Guide, p 147</i>) [must]	X				
o imaging console is located away from radiation source (<i>Guide, p 147</i>)					
<ul style="list-style-type: none"> • Hazardous Agent Containment: NEW 					
o facility adheres to APHIS, USDA and CDC Select Agent Regulations and other federal, state and local regulations including security measures (<i>Guide, p 148</i>) [must]	X				
<ul style="list-style-type: none"> • Behavioral Studies: NEW 					
o facility minimizes airborne transmission of noise and ground-borne transmission of vibration (<i>Guide, p 149</i>)	X				
o floor coverings reduce sound transmission (<i>Guide, p 149</i>)	X				
o testing equipment allows for surface disinfection (<i>Guide, p 150</i>) - one behavior run and one sound chamber had unsealed edges.		X			
o components that cannot be cleaned are not in ready contact with animals and kept covered when not in use (<i>Guide, p 150</i>)	X				
o housing areas are contiguous with testing areas when appropriate (<i>Guide, p 150</i>)	X				

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NA = not applicable

Deficiencies-at-a-Glance by Type
July - Oct 2015

Facility	Room	Degree	Type	Deficiency	Orig. Deadline	Ext. Deadline	Comp. Date	Month
DCM 6th Fl Vivarium	D607	Deficiency	Animal Health	Feed stored in cold room was on the rack against the wall. Feed needs to be stored 6" away from the walls.	8/19/15		8/4/15	July
DCM 6th Fl Vivarium	D607	Deficiency	Animal Health	Expired food present. This was removed during the visit.	Corrected during visit		7/30/15	July
DCM 6th Fl Vivarium	G607	Deficiency	Animal Health	Expired cereal (8/14) was found in drawer. This was discarded during the visit	Corrected during visit		7/30/15	July
DCM Brotman Vivarium	P155	Deficiency	Animal Health	Sunflower seeds in Ziploc bag did not have a use-by or expiration date. This was corrected during the visit.	Corrected during visit		7/27/15	July
DCM Foege Vivarium	N040	Deficiency	Animal Health	Expired Fenbendazole feed with mill date 11/19/14. This was corrected during the visit.	Corrected during visit		7/10/15	July
DCM Hitchcock	118	Deficiency	Animal Health	Wooden perches in bird cages are not sanitizable and must be replaced frequently. Please develop a schedule for replacing wooden perches.	10/30/15		11/9/15	October
DCM Hitchcock	118	Deficiency	Animal Health	Bird seed in the container is without a labeled plastic lid and covered with a blue drape. This was corrected during the visit and the container is covered with a labeled lid.	Corrected during visit		10/2/15	October
DCM Hitchcock	118C	Deficiency	Animal Health	Cuttlefish bones were loose in a drawer and should be stored with the ones in the larger plastic container. This was corrected during the visit.	Corrected during visit		10/2/15	October
DCM Hitchcock	118C	Deficiency	Animal Health	Food in the refrigerator, e.g., greens and eggs, inside their original take-out containers should be transferred to a secondary plastic container. The container should be labeled with the contents, 'not for human consumption', and the use-by-date.	10/30/15		11/9/15	October
DCM HR&T Vivarium	Feed Room	Deficiency	Animal Health	Produce in the refrigerator is not labeled with the purchase date.	8/17/15		Late response 10/19/2015	July
DCM HR&T Vivarium	Feed Room	Deficiency	Animal Health	Mill date and contents are missing on pig feed in barrel. Please label with mill date and contents.	8/17/15		8/17/15	July

Deficiencies-at-a-Glance by Type
July - Oct 2015

DCM Kincaid	558	Deficiency	Animal Health	Feed in plastic storage bin next to the storage racks are labeled with the incorrect mill date. Please ensure that all feed is within its use-by date and that all labels contain relevant dates.	10/28/15		10/6/15	October
DCM Kincaid	558	Deficiency	Animal Health	Blue carboy by sink (probably used for potable water) needs to be labeled with contents.	10/28/15		11/13/15	October
DCM Kincaid	558	Deficiency	Animal Health	Small food pellets needs to be properly labeled. Please label the contents, mill date, and expiration date.	10/28/15		11/13/15	October
DCM Kincaid	558 (Chambers 3B, 5A)	Deficiency	Animal Health	Expired food found in chambers and in plastic storage bin next to the cage setup storage racks. Please dispose of.	10/28/15		10/6/15	October
DCM K-Wing Vivarium	Feed Storage Room	Deficiency	Animal Health	Feed bags were against the wall. Feed needs to be stored 6" away from the walls.	8/7/15		8/3/15	July
DCM K-Wing Vivarium	K018C	Deficiency	Animal Health	Date labeled on food bin suggests food is out of date. At time of visit, Tech filled the two food bins at the same time, but did not date the second bin. Bins must be labeled correctly. This was corrected during the visit.	Corrected during visit		7/8/15	July
DCM SLU 3.1	SB024	Deficiency	Animal Health	One flooded cage was found by site visitors. The cage was changed during the visit.	Immediately		7/22/15	July
DCM SLU 3.1	SB028	Deficiency	Animal Health	Four neonates were found dead by site visitors and were removed and logged appropriately during the visit. (Gathering more information regarding the issue)	Immediately		7/22/15	July
DCM SLU 3.1	SB088E (BSL-2)	Deficiency	Animal Health	Unlabeled and undated food bin contained a food bag that could not be interpreted. The bag was discarded during the visit.	Corrected during visit		7/22/15	July
Guthrie	355	Deficiency	Animal Health	Carboy is labeled as potable water. Please remove the label. All secondary containers must be labeled adequately with contents and expiration date.	11/10/15		11/10/15	October
Guthrie	355	Deficiency	Animal Health	Plastic container with treats must be stored off the floor.	11/10/15		11/6/15	October
Guthrie	410	Deficiency	Animal Health	Carrots in the refrigerator need to be placed in a ziplock bag or plastic container with appropriate labeling. Please label the contents and expiration date.	11/10/15		10/15/15	October
Guthrie	369E	Deficiency	Animal Health	Food bin was uncovered. The lid must be replaced after use.	11/10/15		10/14/15	October

Deficiencies-at-a-Glance by Type
July - Oct 2015

Kincaid	015B	Deficiency	Animal Health	Dead fish need to be stored in a separate freezer from the fish food.	11/9/15		11/9/15	October
SLU	N323	Deficiency	Animal Health	Syringe with sharp needle on the bottom of the clear plastic supply box in the animal use area. Syringe contained fluid and the needle had tubing covering the end. Recommended lab use a blunt needle for fluid delivery device. Lab personnel addressed the issue immediately.	Corrected during visit		8/4/15	August
WaNPRC I-Wing	I363A	Deficiency	Animal Health	Food bag is on the floor and not in a container. Please ensure food is stored appropriately. Food should be stored off the floor and in a sanitizable container to prevent vermin access and soiling.	10/21/15		9/18/15	September
WaNPRC I-Wing	I453	Deficiency	Animal Health	Food is not tagged with an expiration date. The expiration date should be visible on the label so that food is not used past its use by date.	10/21/15		10/9/15	September
WaNPRC I-Wing	I453A	Deficiency	Animal Health	Food (Yoplait) in the refrigerator is without a corresponding label. The facility practice is to provide a label on the front of the refrigerator and this was missing. Please label.	10/21/15		10/9/15	September
WaNPRC I-Wing	I455	Deficiency	Animal Health	Food container is missing a label. Please label with content and an expiration date.	10/21/15		10/9/15	September
WaNPRC I-Wing	I553	Deficiency	Animal Health	4 cans of expired milk and egg protein powder. Please discard.	9/29/15		9/29/15	September
WaNPRC I-Wing	I553	Deficiency	Animal Health	Marshmallows were properly labeled with an open date, but not a use by date. The marshmallows were stored in an open bag that was not secured. Please store marshmallows in a sealed container, bag or box, and label with both open and use by dates.	9/29/15		9/17/15	September
WaNPRC I-Wing	I700T	Deficiency	Animal Health	Apples in the refrigerator is without a corresponding label. The facility practice is to provide a label on the front of the refrigerator and this was missing. Please label.	10/21/15		10/20/15	September
Brotman	327	Deficiency	Drug Storage/Expiration/Documentation	Vials of 'KX' mix were not labeled with the concentration and mix date or expiration date. Please label.	8/6/15		7/28/15	July
CHDD	186	Deficiency	Drug Storage/Expiration/Documentation	Expired heparin was discarded during the visit.	Corrected during visit		7/16/15	July

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CHDD	055	Deficiency	Drug Storage/Expiration/Documentation	Expired gel foam and sutures were donated to AUTS during the visit.	Corrected during visit		7/16/15	July
CHDD	055	Deficiency	Drug Storage/Expiration/Documentation	Expired Ringer's (11/14) was discarded during the visit. Expired cisplatin (5/13) was labeled as expired during the visit.	Corrected during visit		7/16/15	July
CHDD	068	Deficiency	Drug Storage/Expiration/Documentation	Expired sutures were donated to AUTS during the visit.	Corrected during visit		7/16/15	July
DCM 6th Fl Vet Svcs	C603	Deficiency	Drug Storage/Expiration/Documentation	Expired saline was discarded. This was corrected during the visit.	Corrected during visit		7/16/15	July
DCM 6th Fl Vivarium	C608	Deficiency	Drug Storage/Expiration/Documentation	Expired hydrogen peroxide. This was removed during the visit.	Corrected during visit		7/30/15	July
DCM 6th Fl Vivarium	C608	Deficiency	Drug Storage/Expiration/Documentation	Expired Hydrogen Peroxide (3/15). Please dispose of.	8/19/15		Late response 11/6/2015	July
DCM 6th Fl Vivarium	C613	Deficiency	Drug Storage/Expiration/Documentation	Expired Clidox bottle in active behavioral testing cubicle (made 7/10). Please dispose of.	8/19/15		7/31/15	July
DCM 6th Fl Vivarium	C613	Deficiency	Drug Storage/Expiration/Documentation	Expired Clidox. Please dispose of.	8/19/15		7/31/15	July
DCM 6th Fl Vivarium	C615	Deficiency	Drug Storage/Expiration/Documentation	Expired ultrasound gel. Please discard.	8/19/15		Late response 11/6/2015	July
DCM 6th Fl Vivarium	C621	Deficiency	Drug Storage/Expiration/Documentation	Expired PureLube eye gel. Please remove or mark as not to be used in animals.	8/19/15		Lates response 11/6/2015	July
DCM 6th Fl Vivarium	G615	Deficiency	Drug Storage/Expiration/Documentation	Expired Roccal should be disposed of properly.	8/19/15		Late response 11/6/2015	July
DCM 6th Fl Vivarium	G616	Deficiency	Drug Storage/Expiration/Documentation	Expired silver nitrate from 2/2003 was discarded during the visit.	Corrected during visit		7/30/15	July
DCM Brotman Vivarium	C34	Deficiency	Drug Storage/Expiration/Documentation	Expired ketamine in lockbox (exp. 3/15). Buprenorphine diluted in December 2014 is considered expired. Please dispose of properly.	8/18/15		8/3/15	July

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DCM Brotman Vivarium	P152	Deficiency	Drug Storage/Expiration/Documentation	Expired culture swabs in mouse necropsy skits (2/2013 and 3/2012) and in rabbit kits. These were discarded during the visit.	Corrected during visit		7/27/15	July
DCM Brotman Vivarium	P154	Deficiency	Drug Storage/Expiration/Documentation	Expired blood glucose test strips 12/14 (in plastic bin); expired 0.9% sterile saline 7/1/2015 and expired sterile water 11/1/2013. These were removing during the visit.	Corrected during visit		7/27/15	July
DCM Brotman Vivarium	P162	Deficiency	Drug Storage/Expiration/Documentation	Drug log for undiluted buprenorphine did not match what was currently on hand. It appears that a mix was made on 6/29, but was not recorded in the log. This was corrected during the visit.	Corrected during visit		7/27/15	July
DCM Brotman Vivarium	P164	Deficiency	Drug Storage/Expiration/Documentation	Expired cyclosporine tablets 10/8/14 and Surgilube 3/2014. These were discarded during the visit.	Corrected during visit		7/27/15	July
DCM Foege Vivarium	Cage Wash	Deficiency	Drug Storage/Expiration/Documentation	Expired pH solution. This was corrected during the visit.	Corrected during visit		7/10/15	July
DCM Foege Vivarium	Colony Management Procedure Room	Deficiency	Drug Storage/Expiration/Documentation	Expired materials. These were corrected during the visit.	Corrected during visit		7/10/15	July
DCM Foege Vivarium	N031	Deficiency	Drug Storage/Expiration/Documentation	Expired Maxon suture dated 4/2009 needs to be removed.	8/12/15		7/13/15	July
DCM Foege Vivarium	N031	Deficiency	Drug Storage/Expiration/Documentation	Expired H2O2 labeled do not use immediately dated 4/15/15 and expired 0.9% NaCl dated 5/15. Please dispose of properly.	8/12/15		7/13/15	July
DCM Foege Vivarium	N046	Deficiency	Drug Storage/Expiration/Documentation	Expired Proline suture dated 1/2008 and surgery blades dated 10/2013 need to be removed.	8/12/15		7/13/15	July
DCM Foege Vivarium	N046	Deficiency	Drug Storage/Expiration/Documentation	Expired Vet bond dated 3/2009. Please dispose of.	8/12/15		7/13/15	July
DCM Foege Vivarium	Surgery Room	Deficiency	Drug Storage/Expiration/Documentation	Betadine and saline in secondary containers are labeled with 4/16/15, but it is unclear if that is the expiration date or the use date. Please label with the expiration date.	8/12/15		7/13/15	July
DCM Foege Vivarium	Surgery Room	Deficiency	Drug Storage/Expiration/Documentation	Large container of saline labeled 0.9% NaCl is dated 5/17/13. Please clarify the labeled date. Expired solution must be disposed of.	8/12/15		7/13/15	July

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DCM Hitchcock	118 (Main area)	Deficiency	Drug Storage/Expiration/Documentation	Expired bottle of Virkon S. This was disposed of during the visit.	Corrected during visit		10/2/15	October
DCM Hitchcock	118C	Deficiency	Drug Storage/Expiration/Documentation	Eppendorf containing liquid (~1 ml) labeled as Metalan drops stored in the refrigerator. The label is difficult to read and is missing an expiration date. Unclear if this was used in animals. Please verify use in protocol if it is used in animals and specify protocol number and use. If not used in animals, please label 'Do not use in live animals.'	10/30/15		11/6/15	October
DCM HR&T Vivarium	SB011	Deficiency	Drug Storage/Expiration/Documentation	Investigator lockboxes in the facility were located in a refrigerator and others on the walls in procedure rooms. The contents in the lockboxes need to be accessible for site visitors.	8/17/15		8/17/15	July
DCM HR&T Vivarium	SB023	Deficiency	Drug Storage/Expiration/Documentation	Expired capillary tubes (6/31/08). Please dispose of.	8/17/15		8/17/15	July
DCM HR&T Vivarium	SB044	Deficiency	Drug Storage/Expiration/Documentation	Bag of Lactated Ringer's opened 6/26/15. Please dispose of as it expires in 24 hours after opening if kept unrefrigerated, or may be kept up to a week if refrigerated.	8/17/15		7/21/15	July
DCM HR&T Vivarium	SB056	Deficiency	Drug Storage/Expiration/Documentation	Expired media (3/15) found in a cabinet. Please discard or label "Not For Animals Use."	8/17/15		8/17/15	July
DCM K-Wing Vivarium	I012Q	Deficiency	Drug Storage/Expiration/Documentation	Clidox change was completed, but date was not recorded. This was corrected during the visit.	Corrected during visit		7/8/15	July
DCM K-Wing Vivarium	I015N	Deficiency	Drug Storage/Expiration/Documentation	Expired Clidox needs to be changed.	8/7/15		7/8/15	July
DCM K-Wing Vivarium	K018F	Deficiency	Drug Storage/Expiration/Documentation	Clidox in secondary container made on 6/22/15 has been in use longer than 14 days according to the label.	8/7/15		7/8/15	July
DCM SLU 3.1	SB063	Deficiency	Drug Storage/Expiration/Documentation	Tamoxifen and oil were not labeled with an expiration date. The oil was discarded during the visit, but the tamoxifen needs to be labeled appropriately.	8/31/15		8/31/15	July
DCM SLU 3.1	SB084	Deficiency	Drug Storage/Expiration/Documentation	Clidox bottle has a hole which is leaking liquid. Please replace bottle.	8/31/15		7/22/15	July

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DCM SLU 3.1	SB087A	Deficiency	Drug Storage/Expiration/Documentation	Clidox residue is excessive and needs to be wiped off after spraying.	8/31/15		7/22/15	July
DCM T-Wing Vivarium	T125	Deficiency	Drug Storage/Expiration/Documentation	Log for Clidox was missing the year. Please label the dates in this format (MM/DD/YY).	8/12/15		8/12/15	July
Fb029a	B016B	Deficiency	Drug Storage/Expiration/Documentation	Expired Clidox was discarded during the visit.	Corrected during visit		7/17/15	July
Foege	N433A	Deficiency	Drug Storage/Expiration/Documentation	70% EtOH is used to decontaminate instruments and surfaces that BSL-2 mice administered human source material are used in/on. Please contact the manufacturer for recommendations regarding an EPA approved disinfectant (e.g., Clidox).	8/5/15		7/23/15	July
Foege	N433A	Deficiency	Drug Storage/Expiration/Documentation	Expired and unlabeled bupivacaine was removed. This was corrected during the visit.	Corrected during visit		7/7/15	July
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Drug Storage/Expiration/Documentation	Containers with dyes for use with flumes in the lab must be labeled with the contents.	11/13/15		11/6/15	September
Friday Harbor Labs	Lab 8	Deficiency	Drug Storage/Expiration/Documentation	Expired potassium bromide in the refrigerator. This was discarded during the visit.	Corrected during visit		9/16/15	September
Guthrie	410	Deficiency	Drug Storage/Expiration/Documentation	Metacam in 1 ml syringe for oral dosing has inadequate label. The label must include concentration and expiration date.	11/10/15		10/14/15	October
Guthrie	410	Deficiency	Drug Storage/Expiration/Documentation	Evan's Blue in Eppendorf is missing the expiration date. This is made from powder in sterile saline. It cannot be filter sterilized because the dye does not go through the filter.	11/10/15		10/20/15	October
Guthrie	410	Deficiency	Drug Storage/Expiration/Documentation	Expired bone wax, petroleum jelly, Vetbond, epinephrine pellets, sterile flexigrids, dopram. Expired items should be appropriately disposed of or labeled as 'Do not use in live animals.'	11/10/15		10/14/15	October
Guthrie	410	Deficiency	Drug Storage/Expiration/Documentation	Fluid bag labels should include an opening date and an expiration date. Please contact Vet Services to get correct details regarding an expiration date. Please report on the outcome.	11/10/15		10/14/15	October

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Guthrie	052	Deficiency	Drug Storage/Expiration/Documentation	Buprenorphine in a secondary container is not labeled with the concentration. Please label the concentration and mix date or expiration date.	11/10/15		11/6/15	October
Guthrie	052	Deficiency	Drug Storage/Expiration/Documentation	Bottle of 100 ml Beuthanasia used has a discrepancy between what is used and the balance indicated in the log (228.4 ml). Please investigate and correct.	11/10/15		11/6/15	October
HR&T	718	Deficiency	Drug Storage/Expiration/Documentation	Non-pharmaceutical grade ketoprofen was discarded during visit.	Corrected during visit		7/17/15	July
HR&T	320D	Deficiency	Drug Storage/Expiration/Documentation	Expired puralube. This was corrected during the visit.	Corrected during visit		7/17/15	July
HSB	BB628	Deficiency	Drug Storage/Expiration/Documentation	One recently expired drug was labeled and moved to the expired container. This was corrected during the visit.	Corrected during visit		10/15/15	October
HSB	D418	Deficiency	Drug Storage/Expiration/Documentation	Expired 0.9% NaCl. This was corrected during the visit.	Corrected during visit		10/8/15	October
HSB	D626	Deficiency	Drug Storage/Expiration/Documentation	Unlabeled spray bottle needs to be properly labeled so contents are identified.	9/29/15		9/1/15	September
HSB	E064A	Deficiency	Drug Storage/Expiration/Documentation	Expired MS-222 in refrigerator. This was disposed of during the visit.	Corrected during visit		10/8/15	October
HSB	E064A, E064B	Deficiency	Drug Storage/Expiration/Documentation	Rooms have multiple conical vials and containers with vials that are not clearly labeled with the contents/expiration dates. There is metformin in the refrigerator that the manufacturer labels as needing to be used within 12 months of receipt, but it is not labeled with the receipt or expiration dates. Lab member was unsure of its use/purpose at the time of the visit and is checking into this issue/deficiency.	11/6/15		10/9/15	October
HSB	F404, F411	Deficiency	Drug Storage/Expiration/Documentation	Controlled substances are currently stored in a flammables cabinet under a fume hood. If controlled substances will be ordered, a more secure storage system will be required.	11/9/15		10/12/15	October
HSB	F404, F411	Deficiency	Drug Storage/Expiration/Documentation	Expired saline. This was corrected during the visit.	Corrected during visit		10/9/15	October

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HSB	F519	Deficiency	Drug Storage/Expiration/Documentation	Two containers are not labeled. This was corrected during the visit.	Corrected during visit		10/1/15	October
HSB	I201A	Deficiency	Drug Storage/Expiration/Documentation	Plastic box of infusion sets and syringes contain some expired infusion sets. Unclear if these are ever used in live animals. Please clarify and either dispose of or label as 'Expired, do not use in live animals.'	11/10/15		10/20/15	October
HSB	J611L	Deficiency	Drug Storage/Expiration/Documentation	Bottle of sterile saline (500 ml) is being reused and aliquots are in secondary containers (ependorfs). Please label with the expiration date.	11/9/15		11/9/15	October
HSB	RR535	Deficiency	Drug Storage/Expiration/Documentation	Unlabeled jar of clear liquid in the fume hood. Please label.	9/8/15		8/18/15	August
Kincaid	146C	Deficiency	Drug Storage/Expiration/Documentation	Expired drugs found. Please dispose of properly.	10/28/15		10/6/15	October
Kincaid	146C	Deficiency	Drug Storage/Expiration/Documentation	SR-Buprenorphine was not in the lab, however, according to the drug log, SR Buprenorphine should have been present in the drug safe. Group will review the surgical records and correct the drug logs.	10/28/15		10/12/15	October
Pack Forest	Pack Forest	Deficiency	Drug Storage/Expiration/Documentation	Ear cleaner (not in use) is in a secondary container without an expiration date. This was corrected during the visit.	Corrected during visit		10/7/15	October
PBB	162	Deficiency	Drug Storage/Expiration/Documentation	Multiple 50 ml conical tubes containing discolored white/brown powder labeled with MS-222. There was also an unlabeled 50 ml conical of white powder. According to the contact, this is pre-weighed MS-222 powder for anesthetic water baths. Proper labeling for a secondary container of anesthetic would include name of agent, concentration or amount of drug, initials, and an expiration or use-by date as appropriate. The drug should be stored in a similar manner as the parent container of drug or disposed of if not being used (the powder was discolored brown).	9/18/15		8/20/15	August

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PBB	PBB	Deficiency	Drug Storage/Expiration/Documentation	There was a bottle of oxygen test lab that was expired 7/2011. This was removed at the time of the site visit. Please ensure all supplies that may be used on animals or are a component of maintaining/verifying water quality are within date.	Immediately		8/27/15	August
SLU	N101	Deficiency	Drug Storage/Expiration/Documentation	Multiple surface disinfectants are expired and should be disposed of (Coverage Spray TB, chlorhexidine, dilute bleach).	9/4/15		9/4/15	August
SLU	N101	Deficiency	Drug Storage/Expiration/Documentation	Bottle of isoflurane and a conical with acetone is being stored on their sides in a supplies box. Chemicals must be stored upright as the bottles could leak.	9/4/15		9/4/15	August
SLU	N101	Deficiency	Drug Storage/Expiration/Documentation	Expired Vetbond in the supplies box. Please dispose of.	9/4/15		9/4/15	August
SLU	N115	Deficiency	Drug Storage/Expiration/Documentation	Expired Nembutal. This was marked as "Expired" and "Do Not Use" at the time of the visit. Expired drug was not used on animals.	Corrected during visit		8/7/15	August
SLU	N115	Deficiency	Drug Storage/Expiration/Documentation	Expired artificial tears. Also a 50 conical tube containing a clear fluid was not labeled clearly, that may have been labeled "K+X expires 1/27/14. It is not clear what this solution was, but it seems likely that it was expired diluted ketamine/xylazine solution. It was labeled "Do Not Use" at the time of the visit. If it is anesthesia solution, it was expired and the protocol liaison should discuss with the group the proper way to prepare and store diluted drugs (label clearly with drugs, concentrations, date made, date expired; prepare in a sterile vial; generally diluted drugs expire one month from date diluted unless parent drug(s) expire sooner).	9/4/15		9/4/15	August
SLU	N115	Deficiency	Drug Storage/Expiration/Documentation	Expired betadine. This was disposed of during the visit.	Corrected during visit		8/7/15	August
SLU	N217	Deficiency	Drug Storage/Expiration/Documentation	Expired Isoflurane in lockbox. Please dispose of.	8/20/15		8/20/15	August

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SLU	N323	Deficiency	Drug Storage/Expiration/Documentation	Expired suture in surgery area. This was disposed of during the visit.	Corrected during visit		8/4/15	August
SLU	N527	Deficiency	Drug Storage/Expiration/Documentation	Expired surgical gloves. Please dispose of.	8/20/15		8/25/15	August
SLU	S462	Deficiency	Drug Storage/Expiration/Documentation	Drug log contained incomplete entries in regards to calculated volumes and weights. Please contact Gregory Lang of the Washington State Department of Health for assistance.	9/30/15		9/15/15	August
SLU	S485	Deficiency	Drug Storage/Expiration/Documentation	Expired drugs found. The drugs were labeled during the visit.	Corrected during visit		8/24/15	August
SLU 3.1	E420A	Deficiency	Drug Storage/Expiration/Documentation	Surface disinfectant Conflikt is expired. This was disposed of during the visit.	Corrected during visit		8/11/15	August
SLU 3.1	E685	Deficiency	Drug Storage/Expiration/Documentation	Expired blood collection needles were marked "Expired, Do Not Use" during the visit.	Corrected during visit		8/5/15	August
SLU 3.1	E685	Deficiency	Drug Storage/Expiration/Documentation	Expired saline, ketamine, and xylazine were labeled "Expired, Do Not Use" during the visit.	Corrected during visit		8/5/15	August
WaNPRC Arizona	204	Deficiency	Drug Storage/Expiration/Documentation	Sentry safe is not bolted down. Site visitors and staff agreed that it would be best to bolt it to a wall or cabinet for addition security	11/2/15		10/8/15	September
WaNPRC Arizona	T27K (Procedure room)	Deficiency	Drug Storage/Expiration/Documentation	Sperule vaccine is only labeled with the drug name and needs to include an expiration date.	10/19/15		10/17/15	September
WaNPRC Arizona	OR 202	Deficiency	Drug Storage/Expiration/Documentation	Expired sterile water for irrigation. This was disposed of during the visit.	Corrected during visit		9/28/15	September
WaNPRC HSB Lab	I013, I021	Deficiency	Drug Storage/Expiration/Documentation	Expired eye lube. This was disposed during the visit.	Corrected during visit		9/9/15	September
WaNPRC HSB Lab	I013, I021	Deficiency	Drug Storage/Expiration/Documentation	Expired Heparin flush syringes. Bag of saline with added Norcuron has not been used in over a year and should not be reused. Please dispose of appropriately or label as 'Expired, Do Not Use In Animals.'	10/12/15		10/6/15	September

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WaNPRC HSB Lab	I021	Deficiency	Drug Storage/Expiration/Documentation	Bottle of Chlorprothixen HCl was not pharmaceutical grade and unclear if used in animals. Please mark 'Do Not Use In Animals.'	10/12/15		10/6/15	September
WaNPRC I-Wing	I089D	Deficiency	Drug Storage/Expiration/Documentation	Expired Telazol is not labeled as 'Expired Do Not Use.' Please label and dispose of appropriately.	10/21/15		9/11/15	September
WaNPRC I-Wing	I445A	Deficiency	Drug Storage/Expiration/Documentation	Expired items, mostly surgical supplies, such as drapes, gel foam, and 3-way stoppers. Expired items should be discarded or very clearly marked as "Expired, Not For Animal Use.	9/29/15		9/29/15	September
WaNPRC I-Wing	I455	Deficiency	Drug Storage/Expiration/Documentation	Expired sterile water is not labeled as expired. This was corrected during the visit.	Corrected during visit		9/9/15	September
WaNPRC I-Wing	I726	Deficiency	Drug Storage/Expiration/Documentation	Fluid bag was labeled with an open date, but not a use by date. This was corrected during the visit.	Corrected during visit		9/4/15	September
WaNPRC I-Wing	I735	Deficiency	Drug Storage/Expiration/Documentation	Expired drugs were not marked as expired. Please label and dispose of appropriately.	10/21/15		9/11/15	September
WaNPRC RR-Wing	RR-Wing	Deficiency	Drug Storage/Expiration/Documentation	Discrepancy noted in Telazol log. The mistake was discovered and corrected during the visit.	Corrected during visit		9/18/15	September
HR&T	320D	Deficiency	Environment	Temperature range is too low (63-68 degrees; Guide range is 68-72 degrees) and varies by more than 4 degrees within a 24 hour period. Sensaphone alarms were not received by lab members. Temperature swings of greater than 7 degrees were recorded in 4/2015, but not reported to the Facility Manager. Please have HMC Engineering check HVAC. Gaps in door allow light to penetrate to animal cage. Please ensure temperatures are within range notify Veterinary Services if there are any health issues as a result.	Immediately		9/30/15	July
HSB	D435	Deficiency	Environment	Housing room had a very strong mouse smell. Possible issue with ventilation.	11/15/15		11/15/15	October
WaNPRC Arizona	152	Deficiency	Environment	Room was evaluated and is dark in the back corner. The LED light system has been ordered.	11/2/15		10/30/15	September

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WaNPRC RR-Wing	Feed and Bedding Room	Deficiency	Environment	Room temperature was > 71° this week and the Supervisor was not notified. Please develop a plan for keeping the feed and bedding space below 70°.	11/9/15		11/10/15	September
WaNPRC RR-Wing	RR056	Deficiency	Environment	Fan is being used to provide white noise. A noise generator can be used to limit the use of fans in the ABSL-2 areas. Please consult with EH&S if necessary.	10/26/15		9/21/15	September
CHDD	055, 186H	Deficiency	Equipment	Rusty scissors are not sanitizable and needs to be cleaned and sharpened prior to euthanasia.	8/31/15		7/30/15	July
DCM 6th Fl Vivarium	C609	Deficiency	Equipment	Mislabeled container of autoclaved instruments. This was labeled correctly during the visit.	Corrected during visit		7/30/15	July
DCM 6th Fl Vivarium	G609	Deficiency	Equipment	Rusty scraper needs to be removed or replaced.	8/19/15		7/31/15	July
DCM Brotman Vivarium	P155, P166	Deficiency	Equipment	Fire extinguisher is not being checked routinely. Appeared to be last checked in 2008.	9/30/15		9/30/15	July
DCM Brotman Vivarium	P169	Deficiency	Equipment	Carbon dioxide tank was present in the animal room for euthanasia during a potential Mouse Parvovirus outbreak, but there was no signage to indicate the appropriate flow rate to meet current AVMA Euthanasia Guidelines.	8/18/15		8/3/15	July
DCM Foege Vivarium	N048	Deficiency	Equipment	Please label the ice machine as not potable.	8/12/15		7/13/15	July
DCM Foege Vivarium	N064	Deficiency	Equipment	Rusty scissors need to be removed or replaced.	8/12/15		7/13/15	July
DCM Kincaid	558	Deficiency	Equipment	Large red-light lamp needs to be labeled and the smaller red-light flashlights (other handheld, portable red-lights) should have working batteries.	10/28/15		11/13/15	October
Guthrie	028D	Deficiency	Equipment	Rusty instruments for survival surgery must be removed.	10/23/15		10/15/15	October
HR&T	320D	Deficiency	Equipment	Guillotine is rusty, loose, and dirty. Please refer to the guillotine maintenance policy.	8/7/15		9/1/15	July
HSB	H514	Deficiency	Equipment	Isoflurane vaporizer is due for maintenance. In general, servicing is required annually. Please post a sign stating "Do Not Use Until Calibrated." Please contact the vendor to determine the next service date and notify the IACUC.	11/2/15		10/16/15	September

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HSB	I201B	Deficiency	Equipment	Rusty equipment is not sanitizable and should be replaced or removed.	11/10/15		10/20/15	October
HSB	J661A	Deficiency	Equipment	Rust on base of hood. Mold behind hood. Please remove rust and mold.	11/30/15		11/13/15	October
Kincaid	Food Storage Incubator in 558	Deficiency	Equipment	Food storage incubator is not able to keep contents cool due rising to room temperature, which is higher than the 70 degree limit for rodent chow. Please replace or repair incubator, or move the incubator to a cold room or to a cooler location.	11/6/15		11/16/15	October
WaNPRC Arizona	Necropsy	Deficiency	Equipment	Garden shears for bone cutting were rusty. Please replace when rusty and consider stainless steel shears.	10/19/15		10/1/15	September
WaNPRC HSB Lab	I013, I021	Deficiency	Equipment	Vaporizer is due for maintenance. In general, servicing is required annually. Please post a sign stating "Do Not Use Until Calibrated." Please contact the vendor to determine the next service date and notify the IACUC.	10/31/15		10/8/15	September
WaNPRC RR-Wing	RR055	Deficiency	Equipment	Testing apparatus has worn edges and damaged areas. The apparatus needs to be repaired and sealed to be sanitizable.	11/9/15		11/9/15	September
Big Beef Creek Field Station	Rearing Barn (Building #7)	Deficiency	Facility	Damaged ceiling insulation in three different areas within the facility need to be repaired.	2/29/16			August
CHDD	055	Deficiency	Facility	Ceiling tiles, vents, fire sprinklers, and floor drain are dirty and need to be cleaned or replaced.	9/30/15		8/25/15	July
CHDD	056D	Deficiency	Facility	Dirty fire sprinklers need to be cleaned, and the light cover hanging down needs to be reseated.	9/30/15		8/25/15	July
CHDD	072	Deficiency	Facility	Prevalent mouse odors are detectable, likely due to the vivarium next door. This poses an occupational health hazard to lab personnel. Facilities Services will be contacted to test air flow and ventilation system.	8/30/15		8/14/15	July
CHDD	072	Deficiency	Facility	Fire extinguisher has not been serviced since 2013.	9/30/15		9/4/15	July
CHDD	186H	Deficiency	Facility	Door sweep is not present. Animal use locations require a door sweep to prevent animals from escaping.	8/30/15		8/17/15	July

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CHDD	186H	Deficiency	Facility	Ceiling tiles need to be resealed. Exposed drywall next to sink is not sanitizable and needs to be painted.	9/15/15		8/26/15	July
DCM 6th Fl Vet Svcs	A606A	Deficiency	Facility	Cover on electrical panel for alarm is missing. Please replace. OAW will submit a Work Order and Facilities Services will contact to schedule the repair.	9/30/15		8/31/15	July
DCM 6th Fl Vet Svcs	C604	Deficiency	Facility	Holes in the back wall and around the outlet need to be repaired. Rusty plumbing fixtures in the left wall by the floor need to be painted. Area around the thermostat needs to be patched and painted. Holes in the right wall near the sink need to be patched.	9/30/15		9/8/15	July
DCM 6th Fl Vet Svcs	C604	Deficiency	Facility	Seal gap where cabinet has pulled slightly away from the wall.	8/30/15		8/17/15	July
DCM 6th Fl Vet Svcs	C604A	Deficiency	Facility	Repair or replace the molding that has fallen away from the base of the cabinet near the floor.	9/30/15		9/30/15	July
DCM 6th Fl Vet Svcs	C606	Deficiency	Facility	Damaged walls throughout the room need to be repaired.	8/30/15		8/14/15	July
DCM 6th Fl Vivarium	AA702	Deficiency	Facility	Hole under the sink where the drain pipes goes into the wall needs to be sealed to prevent vermin from entering and/or escaping.	9/30/15		8/14/15	July
DCM 6th Fl Vivarium	E612	Deficiency	Facility	Floor drain missing a lid. This is a safety hazard and an access point for vermin.	9/30/15		9/4/15	July
DCM Brotman Vivarium	A39	Deficiency	Facility	Damage on wall next to clock needs painting.	9/30/15		9/4/15	July
DCM Brotman Vivarium	B36 (Dirty Washroom)	Deficiency	Facility	Paint is peeling on back wall coving, around hose reel and over the dump station.	9/30/15	11/30/15		July
DCM Brotman Vivarium	B36 (Dirty Washroom)	Deficiency	Facility	Floor in center of room needs to be repaired.	9/30/15		11/10/15	July
DCM Brotman Vivarium	C37	Deficiency	Facility	Paint in room needs a touch up prior to housing animals.	9/30/15		9/30/15	July
DCM Brotman Vivarium	P154A, P164B, P167, P168, P169	Deficiency	Facility	Paint chips in multiple areas in the rooms require repainting.	9/30/15		9/4/15	July
DCM Brotman Vivarium	P154B, P167A	Deficiency	Facility	Cracked paint on ceiling and around thimbles. Please patch and paint.	9/30/15	11/30/15		July
DCM Brotman Vivarium	P165	Deficiency	Facility	Holes in wall where freezer is located need to be repaired.	9/30/15		9/4/15	July
DCM Brotman Vivarium	P170	Deficiency	Facility	Outlet cover on the left wall is missing and the outlet has been slightly pulled out of the wall.	9/30/15		11/13/15	July

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DCM Foege Vivarium	Feed/Bed Room	Deficiency	Facility	Small wall defects in feed and bedding room need to be repaired.	8/30/15		8/12/15	July
DCM Foege Vivarium	N062	Deficiency	Facility	Drain behind autoclave was not covered by a grater. Please cover.	9/30/15		9/28/15	July
DCM Foege Vivarium	N80A	Deficiency	Facility	Doors in ante room between sterile and clean holding need to be painted.	9/30/15	10/30/15	10/26/15	July
DCM Hitchcock	118A, 118B, 118E	Deficiency	Facility	Exhaust vent in the wall under the sink is dusty. Please clean.	11/6/15		11/5/15	October
DCM HR&T Vivarium	SB074	Deficiency	Facility	Animal room used as storage space has shavings in drain and needs to be cleaned. Another drain is missing a cover, and this presents a tripping hazard. Mops must be hung up off the floor.	8/17/15		7/28/15	July
DCM HSB	J027	Deficiency	Facility	Paint chips on south wall need to be patched and painted.	11/15/15		10/28/15	September
DCM K-Wing Vivarium	I012A	Deficiency	Facility	Rusty floor cleanout cover in back corner needs to be refinished or replaced.	9/15/15		8/26/15	July
DCM K-Wing Vivarium	I015M	Deficiency	Facility	Gap around ceiling fixture needs to be re-caulked, or the caulking removed and the area repainted as needed. Please evaluate all fixtures in the I015 suite for this potential defect.	12/31/15		10/30/15	July
DCM Roosevelt	253	Deficiency	Facility	Rusty shelf bracket above sink cannot be adequately sanitized. Bottom corner of sink cabinet has exposed wood that cannot be sanitized. The cabinet should be replaced. DCM will submit a Work Order to Roosevelt Facility Services.	9/30/15		9/30/15	July
DCM SLU 3.1	SB088B	Deficiency	Facility	BSL-2 room door viewing window has a 2.5-3.5 inch crack and needs to be replaced.	9/30/15	11/30/15		July
DCM T-Wing Vivarium	T119	Deficiency	Facility	Holes and dents in wall need to be repaired.	9/30/15		9/2/15	July
Friday Harbor Labs	Fernald F-116	Deficiency	Facility	Replace stand for sink located in room. Amount of rust on stand is significant enough to potentially be a structural problem in the future in addition to preventing adequate sanitation.	12/31/15		12/16/15	September
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Facility	Eyewash post base is rusty. Please remove rust and repaint or replace.	12/31/15		10/30/15	September
Friday Harbor Labs	Lab 7 (Outdoor tanks)	Deficiency	Facility	One of the two present gray tank lids present will need to be resurfaced with epoxy/urethane paint and the missing third tank lid will need to be replaced before it is used to house vertebrates in summer 2016.	3/31/16		10/30/15	September

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Guthrie	355	Deficiency	Facility	Nail hole in the wall besides the door needs to be patched and painted.	11/15/15		11/16/15	October
Guthrie	421	Deficiency	Facility	Ceiling tiles need to be replaced.	11/15/15		11/6/15	October
Guthrie	044A, 056	Deficiency	Facility	Paint chips in the wall need to be patched and painted.	11/15/15		11/16/15	October
Guthrie	050	Deficiency	Facility	Seal penetration in ceiling around air duct.	11/15/15		10/20/15	October
Guthrie	056	Deficiency	Facility	Old cabinet is not sanitizable and needs to be removed. The old pipes also need to be removed.	2/15/16		FS to provide	October
Guthrie	342B	Deficiency	Facility	Cardboard being used a ceiling tile needs to be removed and replaced with a ceiling tile.	11/15/15		10/27/15	October
Guthrie	351C	Deficiency	Facility	Behavior run has an edge that is unsealed (bottom, short edge).	11/15/15		11/10/15	October
Guthrie	363A	Deficiency	Facility	Seal penetration in ceiling by commutator.	11/15/15		10/20/15	October
Guthrie	Far West Aviary	Deficiency	Facility	Cage with the drain cover pulled up partially. This could present a hazard to the birds in terms of sharp edges and allowing birds access to the drain.	11/15/15		11/9/15	October
Harborview 9th & Jefferson Building	3NJ383.2	Deficiency	Facility	Broken outlet cover needs to be replaced. Group will submit a Work Order to Harborview Engineering.	9/30/15		7/30/15	July
Hitchcock	144A	Deficiency	Facility	Ceiling tile above animal work area needs to be replaced. Several damaged tiles and a few stained/damaged tiles should also be replaced.	11/15/15		11/2/15	October
HR&T	320D	Deficiency	Facility	Door is not secured and also allows light to penetrate.	9/30/15		9/30/15	July
HR&T	320D	Deficiency	Facility	Wall near CO2 has gouges in drywall and is not sanitizable. Holes around the hood could pose a vermin access point.	9/30/15		9/30/15	July
HR&T	B004	Deficiency	Facility	Holes in ceiling tiles above the surgical area. Tiles need to be replaced with ones that can be cleaned. Group will submit a Work Order to Harborview Engineering.	9/30/15		7/16/15	July
HR&T	B016	Deficiency	Facility	Water damaged ceiling tile needs to be replaced and the leak needs to be fixed. Group will submit a Work Order to Harborview Engineering.	9/30/15		8/31/15	July
HR&T	B018C, B018B	Deficiency	Facility	Holes in ceiling tiles. Ceiling tiles need to be replaced. Group will submit a Work Order to Harborview Engineering.	9/30/15		8/24/15	July
HSB	B529	Deficiency	Facility	Broken plastic face plate on data outlet in the back of the room needs to be replaced.	9/30/15		8/31/15	August

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HSB	BB1132	Deficiency	Facility	Rusty front sill on fume hood needs to be repainted.	11/15/15		11/5/15	October
HSB	BB1146	Deficiency	Facility	Exposed pressboard edges on the sound chamber needs to be sealed and painted.	11/15/15		11/6/15	October
HSB	BB1544	Deficiency	Facility	Ceiling tile removed by Facility Services needs to be put back in place by Facility Services.	11/15/15		11/6/15	October
HSB	BB857A	Deficiency	Facility	Non-functional eyewash in lab needs to be repaired.	11/15/15		11/2/15	October
HSB	H225	Deficiency	Facility	Countertop is pitted, and scratched, and no longer sanitizable. Facility Services will evaluate the surface for repair or replacement.	2/28/16		FS to provide	October
HSB	H428B (Surgery room)	Deficiency	Facility	Brick blocks to elevate surgery table are not sanitizable and need to be sealed.	11/30/15		11/16/15	October
HSB	H428B (Surgery room)	Deficiency	Facility	Surgery table extender to lift surgical field is wood and unsealed in some areas. The wood needs to be sealed for a sanitizable surface.	11/30/15		11/16/15	October
HSB	I534T	Deficiency	Facility	Faucet sink is dripping and needs to be repaired. Facilities Services will be contacted to the schedule repair.	10/15/15		9/29/15	September
HSB	J611L	Deficiency	Facility	Wooden drawer unit under the table has raw wood chips that are not sanitizable. The wood needs to be sealed.	11/15/15		11/9/15	October
HSB	J661A	Deficiency	Facility	Ceiling tiles are damaged and need to be replaced.	11/15/15		11/6/15	October
HSB	K127	Deficiency	Facility	Table where animal cages are placed prior to perfusion has holes exposing unsealed wood.	10/30/15		10/5/15	September
HSB	K544	Deficiency	Facility	Ceiling tiles and vent are dirty and need to be cleaned or replaced.	10/15/15		10/5/15	September
HSB	K544	Deficiency	Facility	Countertop is rusted/non-sanitizable and needs to be replaced or resealed.	11/15/15		11/2/15	September
HSB	RR013	Deficiency	Facility	Edge of table has broken off and needs to be laminated.	10/15/15		9/30/15	September
Kincaid	011	Deficiency	Facility	Drain/trap in the floor needs to be cleared of debris. OAW will submit a Work Order and Facilities Services will contact to schedule the repair.	11/15/15		10/29/15	October
Kincaid	Roof Aviaries	Deficiency	Facility	Small strip of sealant in front of cages need to be evaluated by Facilities Services to determine if black mold can be cleaned or removed.	11/15/15		11/16/15	October

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Manchester	Building 12 Incubation Room	Deficiency	Facility	5 tank stands are constructed of unsealed wood. The wood needs to be sealed/painted prior to use.	Prior to use		9/24/15	August
Newport Facility, Newport, OR	Newport Facility	Deficiency	Facility	Cracks in the concrete on the back wall above the freezer need to be repaired and painted to form a sanitizable surface.	3/31/16		3/31/16	October
Newport Facility, Newport, OR	Newport Facility	Deficiency	Facility	Metal poles supporting the facility are rusted at the base. The rust needs to be removed and the area repainted to form a sanitizable surface.	3/31/16		3/31/16	October
PBB	102N	Deficiency	Facility	Floor tiles near the door are badly damaged and need to be replaced.	10/15/15		10/15/15	August
SLU 3.1	E517	Deficiency	Facility	Damage on wall to the right of the door needs to be repainted.	10/30/15	12/30/15		August
WaNPRC Arizona	231	Deficiency	Facility	Rusty hinges are an ongoing issue even though it has been addressed before. The hinges are getting rusty again.	11/2/15		10/2/15	September
WaNPRC Arizona	112, 122	Deficiency	Facility	Rust on ceiling needs to be removed and painted.	10/26/15		10/14/15	September
WaNPRC Arizona	142 under HVAC	Deficiency	Facility	Staining and water running down the walls due to water leaking from the ceiling above HVAC. A work order is in place for repairs and no animals are currently being housed in that area. When necessary, please clean the stains daily.	11/2/15		10/1/15	September
WaNPRC Arizona	241, 242	Deficiency	Facility	Rust spots around well plates need to be removed and painted.	10/26/15		10/1/15	September
WaNPRC HSB Lab	I005	Deficiency	Facility	Paint on the ceiling needs to be patched. Paint chip near the copper lxit line near the west wall.	11/30/15		10/28/15	September
WaNPRC HSB Lab	I013	Deficiency	Facility	Paint chip on the ceiling near the access box near the door to I021. Unfilled nail holes in the east wall under the electrical strip.	11/30/15		10/28/15	September
WaNPRC I- Wing	Booth I	Deficiency	Facility	Booth I: Laminated sign is flaking and exposing paper. Please re-laminate the sign or remove.	9/29/15		9/25/15	September
WaNPRC I- Wing	I009C	Deficiency	Facility	Outlet is shattered and needs to be replaced. Closed piping needs to be repainted.	11/15/15		10/26/15	September
WaNPRC I- Wing	I031	Deficiency	Facility	Wall needs repair and repainting.	11/15/15		10/22/15	September
WaNPRC I- Wing	I031	Deficiency	Facility	Exposed rusty bolts where covers have fallen off and other existing bolt covers need to be repaired.	11/15/15		10/20/15	September
WaNPRC I- Wing	I033	Deficiency	Facility	Door and door jamb needs repair and repainting.	11/15/15		10/28/15	September

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WaNPRC I-Wing	I045	Deficiency	Facility	Chipped walls painted blue and yellow need repair and repainting.	11/15/15		11/4/15	September
WaNPRC I-Wing	I073, I073A	Deficiency	Facility	Corridors need repair and repainting in a several places.	11/15/15		10/26/15	September
WaNPRC I-Wing	I089C	Deficiency	Facility	Door needs painting or replacing.	11/15/15		10/26/15	September
WaNPRC I-Wing	I089Q	Deficiency	Facility	Wall needs repair and repainting.	1/31/16		FS to provide	September
WaNPRC I-Wing	I089R	Deficiency	Facility	Room and door frame needs repair and repainting.	11/15/15		10/26/15	September
WaNPRC I-Wing	I363	Deficiency	Facility	Window covers are delaminating. Please repair or replace the covers.	10/30/15		10/5/15	September
WaNPRC I-Wing	I363, I563, I567	Deficiency	Facility	Wall needs repair and repainting.	11/15/15		10/26/15	September
WaNPRC I-Wing	I445 (booth)	Deficiency	Facility	Un-laminated strip at base. Group is currently looking into getting replacement molding to cover exposed untreated wood.	10/15/15		9/29/15	September
WaNPRC I-Wing	I459	Deficiency	Facility	Window cover needs replacing and resealing.	10/30/15		10/5/15	September
WaNPRC I-Wing	I567	Deficiency	Facility	Pipe insulation needs to be repaired.	10/30/15		10/19/15	September
WaNPRC I-Wing	I715	Deficiency	Facility	Chipped paint on wall above coat hooks by the door (hallway leading to I717) needs to be repaired.	11/15/15		10/5/15	September
WaNPRC I-Wing	I735	Deficiency	Facility	Several light bulbs are not working and need to be replaced.	11/15/15		10/26/15	September
WaNPRC I-Wing	I737	Deficiency	Facility	Light cover is cracked and has a large hole in it.	11/15/15		10/26/15	September
WaNPRC I-Wing	J Corridor	Deficiency	Facility	Door jambs outside of the cagewash room need repair and repainting.	11/15/15		11/10/15	September
WaNPRC I-Wing	J025A	Deficiency	Facility	Rusty bolts need to be replaced.	11/15/15		10/26/15	September
WaNPRC I-Wing	J025A, J025C	Deficiency	Facility	Wall and ceiling need repair and repainting.	11/15/15		10/26/15	September
WaNPRC I-Wing	J025B	Deficiency	Facility	Light switch is broken and needs to be replaced.	11/15/15		10/26/15	September
WaNPRC Western	109	Deficiency	Facility	Vent needs to be secured to wall.	11/10/15	11/30/15	11/17/15	September
WaNPRC Western	112	Deficiency	Facility	Light socket needs to be repaired.	11/10/15	11/30/15	11/13/15	September

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WaNPRC Western	116	Deficiency	Facility	Vent needs to be replaced.	11/10/15	11/30/15	11/12/15	September
WaNPRC Western	316	Deficiency	Facility	Water line is dripping. This is being repaired today.	10/27/15		9/29/15	September
WaNPRC Western	317	Deficiency	Facility	Sprinkle head needs flange.	11/10/15	11/30/15	11/13/15	September
WaNPRC Western	100G, 113, 106	Deficiency	Facility	Chipped paint needs to be repainted.	12/31/15	3/31/16		September
WaNPRC Western	105D	Deficiency	Facility	Emergency shower cover needs to be reattached.	11/10/15	11/30/15	11/12/15	September
WaNPRC Western	200E corridor	Deficiency	Facility	Examine the cover on the Edstrom pressure reducing stations and see if the cover can be cleaned up.	11/10/15	11/30/15	11/18/15	September
WaNPRC Western	234 (Endoscopy room)	Deficiency	Facility	Floor drain needs a cover.	11/10/15		11/10/15	September
CHDD	056E	Deficiency	Husbandry	Arrival dates for zebrafish larvae were missing. Dates when zebrafish are housed in the incubator need to be clearly documented.	8/14/15		7/30/15	July
CHDD	056E	Deficiency	Husbandry	Zebrafish incubator does not have an associated care log, and temperatures are not being recorded regularly. Sensaphone call list is not present.	8/14/15		7/30/15	July
DCM 6th Fl Vivarium	C613	Deficiency	Husbandry	Wire tops and filters were last changed on 6/12, according to the log. Cage tops should be changed every 4 weeks. Please ensure cage tops are changed at least every 4 weeks and recorded in a log.	8/19/15		Late response 10/30/2015	July
DCM 6th Fl Vivarium	E611	Deficiency	Husbandry	The dirty side of the cagewash is missing log entries for this week because the tech is on vacation and there is no back up. Please ensure there is a back-up plan.	8/19/15		8/5/15	July
DCM 6th Fl Vivarium	G606	Deficiency	Husbandry	Air vents need to be cleaned regularly. Please clean and provide the date when completed and a plan for regular cleaning.	8/19/15		8/3/15	July
DCM 6th Fl Vivarium	G619	Deficiency	Husbandry	Cage (Rack 2, side B) was last changed on 7/7/15. According to the posted schedule, the cage is due for changing and some cages have excessive feces build up. Please ensure cages will be cleaned according to the schedule.	8/19/15		8/4/15	July

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DCM 6th Fl Vivarium	G619	Deficiency	Husbandry	Sentinel cage card did not have all the required information. The card only indicated when the cage was set up. Please include Protocol #, contact information, animal source, date of birth or arrival date.	8/19/15		8/19/15	July
DCM 6th Fl Vivarium	H606	Deficiency	Husbandry	Temperature has fluctuated more than 4 degrees for 2 days as indicated on the daily care log. There is no indication that this has been reported to the supervisor. Please include the temperature range for housing ferrets in the daily care log. According to a sign on the outside of G620, the temperature range should be 40 – 65 degrees. Some temperature recordings were above 65. Please make sure that the room is being maintained in the proper range for the species and that that out of range temperatures and fluctuations greater than 4 degrees are reported to the Facility Supervisor promptly.	8/12/15		8/12/15	July
DCM Brotman Vivarium	A39	Deficiency	Husbandry	Feeding logs for special request feed are not filled out daily as specified on request form. Only the most recent logs are present. Feed logs should remain with the rack for the duration of request.	8/18/15		8/18/15	July
DCM Brotman Vivarium	B52	Deficiency	Husbandry	High temperature in feed room is 70.3°. Please adjust setting to lower the temperature below 70°.	8/18/15		8/17/15	July
DCM Brotman Vivarium	P164B	Deficiency	Husbandry	Rack 39/50 had multiple instances of cages not being changed at 14 days. These were all when day 14 was on a holiday. Please ensure the cages will be changed over the holidays.	8/18/15		8/6/15	July
DCM Brotman Vivarium	P167A	Deficiency	Husbandry	Three cages of rats did not have enrichment items.	8/18/15		7/28/15	July
DCM Brotman Vivarium	P168	Deficiency	Husbandry	Two cages of mice did not have nestlets or other enrichment items.	8/18/15		7/29/15	July
DCM Brotman Vivarium	P168	Deficiency	Husbandry	One cage of sentinels did not have nestlets.	8/18/15		7/29/15	July
DCM Brotman Vivarium	P169	Deficiency	Husbandry	Cage cards on breeder cages did not include the date of birth (DOB) for current litters. PIs are responsible for providing the birth dates of litters in the cage.	8/18/15		7/30/15	July

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DCM Foege Vivarium	N047, N049	Deficiency	Husbandry	Cages change interval was longer than 14 days likely due to the room being empty, but was not documented in the log. Please clarify in the log.	8/12/15		7/13/15	July
DCM Foege Vivarium	N080A	Deficiency	Husbandry	Please drape and cover cages.	8/12/15		7/29/15	July
DCM Hitchcock	118A	Deficiency	Husbandry	One bird is singly housed, however, single housing is not approved in the protocol. Please address this situation by submitting a Significant Change to request single housing or explain the situation.	10/30/15		11/5/15	October
DCM Hitchcock	118A	Deficiency	Husbandry	Cage card is missing the hatch date and/or arrival date. Some cages have the incorrect room number on the cage and one flight of cage did not have cage cards.	10/30/15		11/6/15	October
DCM Hitchcock	118B	Deficiency	Husbandry	Cage card did not include the age of the bird. PIs are responsible for labeling the cage cards.	10/30/15		11/6/15	October
DCM Hitchcock	118C (Store room)	Deficiency	Husbandry	Daily tasks have not been checked on the Daily Care Log for Thursday and Friday. For weekly tasks, the wash/wipe door/door jamb was not checked for one week.	10/30/15		10/30/15	October
DCM HR&T Vivarium	Autoclave log books	Deficiency	Husbandry	One entry had both positive and negative circled, and several entries did not have either circled. This was corrected during the visit.	Corrected during visit		7/1/15	July
DCM HR&T Vivarium	Feed Room	Deficiency	Husbandry	Temperature in feed room is 70.7 degrees. Please adjust setting to lower the temperature below 70 degrees.	8/17/15		8/17/15	July
DCM HR&T Vivarium	SB015	Deficiency	Husbandry	Trio breeding cage containing 14 pups with some 18 days old and others 10 days old. Pups need to be separated per posted instructions for trio breeding.	8/17/15		7/20/15	July
DCM HR&T Vivarium	SB015	Deficiency	Husbandry	Bedding in single housing static cages are being changed only every two weeks. Please confirm it will be changed weekly.	8/17/15		8/17/15	July
DCM HR&T Vivarium	SB019	Deficiency	Husbandry	Date of birth is missing on at least two breeding cages, although furred pups were present inside. Date of birth is inconsistently present on other cages. Please label cages with date of birth.	8/17/15		8/4/15	July
DCM HR&T Vivarium	SB027, SB082	Deficiency	Husbandry	Pair of rats on rack 31 are large. Rats > 500 g need to be housed one per cage. Please weigh rats regularly and record weights so that it is clear if/when they need to be separated.	8/17/15		Solution by 11/30/2015	July

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DCM HR&T Vivarium	SB031	Deficiency	Husbandry	Static rack not sanitized in May, 2015, but was cleaned in April and June. According to DCM SOP, rack needs to be cleaned monthly.	8/17/15		8/17/15	July
DCM HR&T Vivarium	SB044	Deficiency	Husbandry	Rabbit restrainers have fur in them. Restrainers need to be cleaned immediately after use.	8/17/15		8/10/15	July
DCM HR&T Vivarium	SB053	Deficiency	Husbandry	Change schedule on rack 23 has not been updated since May. However, the sentinel cage appears clean, which may suggest cleaning has been performed but not logged.	8/17/15		8/17/15	July
DCM Kincaid	558	Deficiency	Husbandry	Several chambers do not have a clearly posted light cycle. Please label.	10/28/15		11/13/15	October
DCM Kincaid	558	Deficiency	Husbandry	Containers of autoclaved water bottles did not have autoclaved dates labeled. Please label.	10/28/15		10/28/15	October
DCM K-Wing Vivarium	K017F	Deficiency	Husbandry	Two overcrowded cages containing 6 adult mice, 25+ grams, were born in March of 2015. Group has variance to wean with 6 to a cage, but should be separated when animals are 5 - 6 weeks old as stated in current protocol.	8/7/15		8/10/15	July
DCM K-Wing Vivarium	K017F	Deficiency	Husbandry	Weaning variance was posted in 2011 and does not contain appropriate expiration date of protocol (5/21/2017). Please update variance with appropriate expiration date.	8/7/15		8/3/15	July
DCM K-Wing Vivarium	K018C	Deficiency	Husbandry	Sign on the door states there is a protocol variance, but the variance could not be located. Please locate and post the variance on the door.	8/7/15		7/31/15	July
DCM K-Wing Vivarium	K018D	Deficiency	Husbandry	Protocol variance posted on door, but none posted in room.	8/7/15		8/10/15	July
DCM K-Wing Vivarium	K018F	Deficiency	Husbandry	Weaning variance is missing the protocol expiration date for group (10/3/2016) and group (4/11/2016). Please add expiration date to sign.	8/7/15		8/4/15	July
DCM K-Wing Vivarium	K-Wing	Deficiency	Husbandry	Sentinel cage cards are missing the required information: strain, contact information and vendor. Please ensure cage cards are labeled correctly.	8/7/15		7/31/15	July
DCM K-Wing Vivarium	Throughout K-Wing	Deficiency	Husbandry	Monthly logs are not being initialed by the Supervisor when checked.	8/7/15		7/29/15	July

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DCM Northlake Diesel Facility	Northlake Diesel	Deficiency	Husbandry	Cages housing single mice were missing the gender information, also please indicate if mice are pregnant.	9/24/15		Late response, completed by 11/13/2015	August
DCM Roosevelt	254	Deficiency	Husbandry	Several cage cards do not include strain information and multiple breeder cage cards do not have birth dates listed.	8/6/15		8/6/15	July
DCM Roosevelt	254	Deficiency	Husbandry	Cages marked "Do Not Change" are missing associated Special Service Requests (SSRs).	8/6/15		8/6/15	July
DCM SLU 3.1	SB026	Deficiency	Husbandry	Several cage cards do not list birthdates for recent litters of pups, which are necessary to determine the appropriate weaning date. Cage cards must include all pertinent dates.	8/31/15		8/31/15	July
DCM SLU 3.1	SB027, SB082	Deficiency	Husbandry	Weekly rack pre-filter change out for rack 16 (room SB027) and racks 82 and 52 (room SB082) were 2 weeks out of date. Please follow established schedule for rack maintenance.	8/31/15		Late Response 10/29/2015	July
DCM SLU 3.1	SB088C	Deficiency	Husbandry	Cage cards are not identifiable as biohazard cards because they were bleached out rather than being colored orange or red. Please replace these cards with new, easily identifiable cage cards.	8/31/15		8/5/15	July
DCM T-Wing Vivarium	T117	Deficiency	Husbandry	Daily care log for Sunday, 7/19/15 was not complete. Please ensure daily care logs are completed.	8/12/15		7/24/15	July
Friday Harbor Labs	Lab 8	Deficiency	Husbandry	Two tanks did not have tank cards. Lab can either clarify general signage ("all fish in lab are on protocol X unless otherwise posted") or provide new tank cards for large tanks.	Immediately		9/18/15	September
Friday Harbor Labs	Lab 8	Deficiency	Husbandry	No care logs are visibly present for the fish currently being housed. Please clarify the location of the current logs or provide new logs for immediate use. Organize past logs from use earlier in the year for future reference.	Immediately		9/18/15	September
Guthrie	044	Deficiency	Husbandry	Temperature fluctuations were recorded. Please ensure that the room is being maintained in the proper range for the species and that that out of range temperatures and fluctuations greater than 4 degrees are reported to the Facility Supervisor promptly.	11/10/15		11/11/15	October

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Guthrie	044	Deficiency	Husbandry	Some birds are singly housed. Some single housing is post-surgery. This needs to be in the EEC and protocol. Please describe reason for single housing.	11/10/15		11/4/15	October
Guthrie	044 (Housing rooms)	Deficiency	Husbandry	Cage cards only contain information of animal ID and treatments. Need: Protocol #, contact info, animal source, birth or arrival date, strain, birth date(s) of offspring (if any). DCM can provide cage cards.	11/10/15		11/4/15	October
Guthrie	051A	Deficiency	Husbandry	Log does not indicate floor cleaning is scheduled on a regular basis. Please develop a plan and document the cleaning in a log.	11/10/15		11/10/15	October
Guthrie	052	Deficiency	Husbandry	While surgery is taking place, how much activity in the room is limited to the surgical procedure was not clear to site visitors.	11/10/15		11/6/15	October
Guthrie	056	Deficiency	Husbandry	Aviary cages need to be cleaned on a monthly basis.	11/10/15		11/11/15	October
Guthrie	056	Deficiency	Husbandry	Food storage bins need to be up off the floor. A system with wheels is recommended for floor cleaning convenience.	11/10/15		11/16/15	October
Guthrie	369C, 369D	Deficiency	Husbandry	Water bottles are missing an autoclaved date. Please label.	11/10/15		11/10/15	October
Guthrie	369D	Deficiency	Husbandry	Cage change was last logged on 10/1/15 on the daily care log. Please ensure the cages are changed and recorded.	11/10/15		11/10/15	October
Guthrie	369G	Deficiency	Husbandry	Some days show a '0' under the column for feed in the feed log. Unclear if this means that animal was not fed, e.g., had no food in their hopper, or that no food was eaten? Please clarify and update log columns so the meaning is clear.	11/10/15		11/6/15	October
Guthrie	369G	Deficiency	Husbandry	Unable to determine when pups present in cage #2 are due for weaning based on cage card information. Please ensure this information is present.	11/10/15		11/6/15	October
Guthrie	Roof Aviary Cages	Deficiency	Husbandry	Cages are sanitized between batches, however, there is no test for effectiveness.	11/10/15		11/11/15	October
Hitchcock	144A	Deficiency	Husbandry	Animals to be euthanized were temporary stored in a non-functioning refrigerator to keep the animals out of sight. Visitors suggested placing the animals waiting to be euthanized on a cart or bench top and draped.	10/28/15		10/9/15	October

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HSB	B108	Deficiency	Husbandry	From the well-maintained post-surgical records, site visitors identified at least one unapproved anesthetic event on 06/06/2014 to replace sutures that had come out 3 days post-op. As far as visitors could tell, Vet Services was not contacted prior to repairing the sutures. Animal was in good health, and recovery was uneventful.	9/30/15		9/15/15	September
HSB	BB1136	Deficiency	Husbandry	Weekly care logs are missing some lab specific tasks. Tasks are being completed, however, some weekly items on some sheets were not recorded. Lab manager will follow up with husbandry staff to remind them to check and make sure weekly tasks are being recorded.	11/9/15		10/20/15	October
HSB	BB1546	Deficiency	Husbandry	Food and water were present in all cages (stocked water bottles did not have a received & expiration date). There were significant amounts of fecal material in the cages. Lab personnel could not provide evidence that the cage had been changed. Please develop a weekly log and keep logs together in one location.	Immediately		10/23/15	October
HSB	BB1546	Deficiency	Husbandry	An EEOP for surgeries and housing needs to be developed.	10/28/15		10/23/15	October
HSB	BB1546	Deficiency	Husbandry	Stocked water bottles are missing an autoclaved date. Please label.	10/28/15		10/23/15	October
HSB	K561	Deficiency	Husbandry	Care log was not completed for the weekend of 9/12-13. Animals housed in decentralized facilities must be checked daily, including weekends and holidays. In addition, the care check must be documented on the log. Please remind personnel to log all animal care activities.	Immediately		9/14/15	September
HSB	K561	Deficiency	Husbandry	Care logs are not being reviewed weekly by a supervisor. Please have supervisor and/or PI review and sign off on care logs.	Immediately		9/14/15	September
HSB	RR013B	Deficiency	Husbandry	Daily and weekly care logs need to be completely filled out. Group needs to develop an SOP to ensure that all cages are being changed weekly and the work is documented.	9/29/15		9/13/15	September

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Kincaid	603A	Deficiency	Husbandry	Cage cards does not contain the required information e.g., PI, protocol #, species. Information may be reported on a general 'cage card sign' that is posted on the door to the lab.	11/9/15		11/9/15	October
PBB	PBB	Deficiency	Husbandry	Most signage for decentralized housing contact names/numbers were outdated. Please ensure all signs are updated. Please contact OAW for this information.	9/30/15		8/20/15	August
PBB	PBB	Deficiency	Husbandry	There was no signage for reporting animal welfare concerns. Per guidance and institutional requirements, this information should be prominently displayed in animal housing areas so personnel are aware of the possible ways to report a concern. Please contact OAW for this information.	9/30/15		8/20/15	August
PBB	PBB	Deficiency	Husbandry	Decentralized housing binders that include important information such as reporting sick animals, standard operating procedures, contact information, and emergency responses were missing. Please contact OAW for this information.	9/30/15		8/20/15	August
PBB	PBB	Deficiency	Husbandry	Most signage for decentralized housing contact names/numbers were outdated. Please ensure all signs are updated. Please contact OAW for this information.	9/30/15		9/30/15	August
PBB	PBB	Deficiency	Husbandry	There was no signage for reporting animal welfare concerns. Per guidance and institutional requirements, this information should be prominently displayed in animal housing areas so personnel are aware of the possible ways to report a concern. Please contact OAW for this information.	9/30/15		9/30/15	August
PBB	PBB	Deficiency	Husbandry	Decentralized housing binders that include important information such as reporting sick animals, standard operating procedures, contact information, and emergency responses were missing in the majority of the housing rooms. Please contact OAW for this information.	9/30/15		9/30/15	August
WaNPRC Arizona	142, AA103	Deficiency	Husbandry	Check sheets are not sanitizable. Paper must be encased in plastic covers.	10/19/15		9/28/15	September

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WaNPRC Arizona	WaNPRC Arizona	Deficiency	Husbandry	Several entries on the weekly checklist for husbandry were checked marked when should have been initialed.	10/19/15		10/5/15	September
WaNPRC Arizona	WaNPRC Arizona	Deficiency	Husbandry	Log sheet does not clearly show which tasks are daily, weekly, etc. and whether or not have been completed. Please reformat log into daily, weekly, and intermittent categories.	10/19/15		10/12/2015 11/2/2015	September
WaNPRC Arizona	WaNPRC Arizona	Deficiency	Husbandry	Pen washing is currently not being logged.	10/19/15		10/12/15	September
WaNPRC I-Wing	Food Storage Room	Deficiency	Husbandry	Large temperature fluctuations were recorded.	10/21/15		9/14/15	September
WaNPRC I-Wing	I047A	Deficiency	Husbandry	Most dates in the sterilizer log are complete, however, some dates are missing biological testing. Please ensure the testing will be completed daily.	10/21/15		10/2/15	September
WaNPRC I-Wing	I559	Deficiency	Husbandry	Hallway cleaning checklist has not been updated since 6/1/15.	10/21/15		10/13/15	September
Western Fisheries Research Center	D119A	Deficiency	Husbandry	One tank did not have a card labeled. Please ensure the tank cards are present on all tanks.	8/28/15		8/21/15	August
Brotman	225	Deficiency	Occupational Health	Eyewash is not being flushed and logged weekly. Eyewash station requires flushing once per week and must be recorded in a log.	7/28/15		7/15/15	July
Brotman	319	Deficiency	Occupational Health	Uncovered waste containers need to be covered.	8/6/15		7/28/15	July
Brotman	319	Deficiency	Occupational Health	Unsecured waste containers on top of benches need to be secured.	8/31/15		7/28/15	July
Brotman	431	Deficiency	Occupational Health	Eyewash needs to be flushed and logged weekly. Eyewash station requires flushing once per week and must be recorded in a log.	8/4/15		7/9/15	July
Brotman	431	Deficiency	Occupational Health	Recapped needle was found in the biohazard container. Used needles should not be recapped for safety measures. Please remind personnel to not recap needles or to use a Needle Recapper.	8/4/15		7/9/15	July
CHDD	186H	Deficiency	Occupational Health	Coffee cup in lab space indicates beverage consumption with the animal use location. Please post "No Food or Drink" signs. PI must remind lab members that eating and drinking is not allowed in the lab.	8/14/15		7/30/15	July

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DCM 6th Fl Vivarium	C621	Deficiency	Occupational Health	Room is cluttered with wires and equipment on the floor. Please organize and clean area.	8/19/15		Late response 11/6/2015	July
DCM 6th Fl Vivarium	G615, hallway near D607	Deficiency	Occupational Health	Eyewash flushing has not been logged for over a week. G615 has not been logged since April 2015. Eyewash station requires flushing once per week and must be recorded in a log.	8/19/15		8/14/15	July
DCM HR&T Vivarium	SB056	Deficiency	Occupational Health	EH&S evaluated SB056 and determined the lab space is BSL-2 and an eyewash station is required. DCM will submit a Work Order to Harborview Engineering.	10/30/15		WO# submitted 8/10/2015	July
DCM Kincaid	558 (Chamber 2A)	Deficiency	Occupational Health	Hazard sticker (flammable) should be applied to bottles containing ethanol.	10/28/15		11/13/15	October
DCM Kincaid	558 (Chamber 5A)	Deficiency	Occupational Health	Wires on the floor to the left of the door upon entry. Please lift wires up off the floor to allow the floor to be easily cleaned.	10/28/15		11/13/15	October
DCM K-Wing Vivarium	I012G, I015A, I015N, I015M	Deficiency	Occupational Health	Expired hand soap in sinks. This was corrected during the visit.	Corrected during visit		7/8/15	July
DCM SLU 3.1	SB082	Deficiency	Occupational Health	Eyewash has not been logged for more than a week. Eyewash station requires flushing once per week and must be recorded in a log.	8/31/15		8/31/15	July
DCM SLU 3.1	SLU 3.1	Deficiency	Occupational Health	Evacuation route maps were not observed by the site visitors and should be accessible to all personnel, as well as visitors to the facility in case of emergency.	9/30/15		8/20/15	July
Foegen	N415A	Deficiency	Occupational Health	F/AIR canister in use without pre-use weight recorded. Canisters used must be weighed before usage and logged indicating the number of hours used.	7/28/15		7/7/15	July
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Occupational Health	Trash cans in animal housing and procedure areas need lids. Please replace trash cans with plastic trash cans with step-on lids.	11/13/15		11/6/15	September
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Occupational Health	Eyewash log is missing dates of entry. By FHL SOP, all eyewashes are supposed to be logged and flushed weekly.	10/30/15		9/30/15	September
Guthrie	355	Deficiency	Occupational Health	Loose electrical cords are a tripping hazard. Please tie up electrical cords and store away from walkway.	11/10/15		11/6/15	October

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Guthrie	410	Deficiency	Occupational Health	Homemade Eppendorf and rack being used as a recapper is a hazardous risk for personnel. OAW will provide a needle recapper.	11/10/15		11/16/15	October
Guthrie	351C	Deficiency	Occupational Health	Ladder in the work area must be removed and stored in a non-animal use location.	11/10/15		11/6/15	October
HR&T	B029A	Deficiency	Occupational Health	Recapped needle was found in the sharps container and the lab is not using a recapper. Used needles should not be recapped for safety measures. Please remind techs to not recap needles or to use a Needle Recapper. OAW will provide a Needle Recapper.	8/6/15		8/6/15	July
HSB	AA025	Deficiency	Occupational Health	Eyewash station shared with a nearby lab is not within 50 feet walking distance to the lab. Group will begin using the eyewash that is in the lab. Eyewash station requires flushing once per week and must be recorded in a log.	8/12/15		8/12/15	July
HSB	AA048F	Deficiency	Occupational Health	Eyewash is not being flushed and logged weekly. Eyewash station requires flushing once per week and must be recorded in a log.	8/12/15		Late response 9/30/2015	July
HSB	BB1502	Deficiency	Occupational Health	Eyewash log is missing several dates. Eyewash station requires flushing once per week and must be recorded in a log.	10/28/15		10/20/15	October
HSB	D626	Deficiency	Occupational Health	Biohazard container in hood is overfilled. Please empty container and do not allow it to overfill.	9/29/15		9/15/15	September
HSB	F404	Deficiency	Occupational Health	Eyewash is not flushed and logged weekly. Eyewash station requires flushing once per week and must be recorded in a log.	11/9/15		10/12/15	October
HSB	F518, F519	Deficiency	Occupational Health	Eyewash is not being flushed on a regular basis. Eyewash must be flushed once a week and recorded in a log.	10/28/15		10/16/15	October
HSB	F518A	Deficiency	Occupational Health	Eyewash is not being flushed. Eyewash must be flushed once a week and recorded in a log.	10/28/15		10/16/15	October
HSB	H073	Deficiency	Occupational Health	Eyewash is flushed weekly, but the log was not adequate. This was corrected during the visit.	Corrected during visit		10/27/15	October
HSB	J661A	Deficiency	Occupational Health	Repeat: Eyewash is not tested weekly. Eyewash station requires flushing once per week and must be recorded in a log.	11/16/15		11/5/15	October
HSB	K084	Deficiency	Occupational Health	Eyewash log needs to be encased in a sanitizable plastic sleeve.	10/12/15		9/29/15	September

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HSB	K084	Deficiency	Occupational Health	Eyewash has not being flushed and logged for several weeks. Eyewash station requires flushing once per week and must be recorded in a log.	10/12/15		9/29/15	September
SLU	N101	Deficiency	Occupational Health	ABSL-2 door signage needs to be updated. A new orange sheet with a list of agents and responsible needs to be posted. The sheet should be posted when those agents/animals are being used in the room, and removed when they are not.	9/4/15		9/4/15	August
SLU	N117	Deficiency	Occupational Health	Eyewash is not being flushed weekly. Eyewash station requires flushing once per week and must be recorded in a log.	8/20/15		8/4/15	August
SLU 3.1	E517	Deficiency	Occupational Health	Gas canisters were very loosely chained and not secured in the event of an earthquake. This was corrected during the visit.	Corrected during visit		8/24/15	August
WaNPRC Arizona	204	Deficiency	Occupational Health	Ladder was propped up against some shelving. This presents a potential tripping/falling hazard. Please store in a safe manner.	10/19/15		10/1/15	September
WaNPRC I-Wing	I-Wing Vivarium	Deficiency	Occupational Health	For the most part, the eyewash log shows that the eyewash is flushed weekly. However, there are instances, 8/2/15 – 8/16/15 and 4/11/15 – 4/25/15, where flushing was not done weekly. Please provide a plan to ensure the eyewash is flushed weekly.	10/28/15		9/18/15	September
WaNPRC I-Wing	I700U	Deficiency	Occupational Health	BSL-2 trash is overfilled. Please develop a schedule for emptying the bin.	10/28/15		10/8/15	September
WaNPRC Western	Throughout facility	Deficiency	Occupational Health	Eyewash needs to be checked annually.	12/31/15			September
HR&T	418	Significant	Protocol	Post-surgical analgesia is not being administered according to approved protocol. Time between doses is too short or too long, and not being administered for the full 48 hours. Scientific Liaison will contact group regarding potential for adding SR buprenorphine to protocol.	Immediately		8/7/15	July
Brotman	319	Deficiency	Sanitation	Paper taped to fume hood is not sanitizable. Paper must be laminated or encased in plastic.	8/6/15		7/28/15	July
Brotman	P124	Deficiency	Sanitation	Remove all tape in the room and remove cardboard box being used as a perfusion stand in room P124.	7/27/15		7/3/15	July

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Brotman	P125B	Deficiency	Sanitation	Tape labels are unsanitizable. Please remove tape and replace on a regular basis. This was corrected during the site visit.	Corrected during visit		7/2/15	July
CHDD	068, 175	Deficiency	Sanitation	Brooms resting on the floor need to be stored off the ground or moved out of the animal use area.	8/14/15		7/30/15	July
DCM 6th Fl Vivarium	C608	Deficiency	Sanitation	Old tape may leave a residue that is unsanitizable. Please remove tape.	8/19/15		Late response 11/6/2015	July
DCM 6th Fl Vivarium	G609	Deficiency	Sanitation	Exposed paper in bins (enrichment logs) and no cover over care log. Paper is unsanitizable and needs to be encased in plastic.	8/19/15		7/31/15	July
DCM 6th Fl Vivarium	G617	Deficiency	Sanitation	Corkboard squares on the cart are not sanitizable. They need to be stored in an enclosed, sanitizable container, such as plastic. Please describe the use of these and when they are disposed.	8/19/15		Late response 11/3/2015	July
DCM 6th Fl Vivarium	G617	Deficiency	Sanitation	Paper calendar needs to be removed. The CO2 sign that is taped to the wall has become dirty. Please remove tape. Sign is not sanitizable and needs to be laminated or encased in plastic.	8/19/15		8/14/15	July
DCM Brotman Vivarium	B42	Deficiency	Sanitation	Autoclave logs need to be laminated or encased in plastic.	8/18/15		8/18/15	July
DCM Foege Vivarium	Animal Rooms	Deficiency	Sanitation	Dusty ceilings around vents in animal rooms need to be cleaned.	9/30/15		8/3/15	July
DCM Foege Vivarium	Halls	Deficiency	Sanitation	Floors in the hall by the cage wash are dirty and need to be cleaned.	8/12/15		8/8/15	July
DCM Foege Vivarium	N048	Deficiency	Sanitation	Hall and ceiling are dirty and need to be cleaned.	8/12/15		7/31/15	July
DCM Foege Vivarium	N080	Deficiency	Sanitation	Floors in the dirty cage wash and clean cage wash are dirty on the edges. Please clean.	8/12/15		8/8/15	July
DCM Foege Vivarium	N080A	Deficiency	Sanitation	Edges of floors, especially the back wall furthest from the autoclave need to be cleaned.	8/12/15		8/8/15	July
DCM Hitchcock	118	Deficiency	Sanitation	Corners behind unused cage wash are dirty and have dust build up. Please clean and develop a regular cleaning schedule.	10/30/15		11/10/15	October
DCM Hitchcock	118	Deficiency	Sanitation	Residue on the floor by the unused cagewash needs to be removed.	10/30/15		10/30/15	October
DCM Hitchcock	118	Deficiency	Sanitation	Blue tape used to post signage above the sink area in the main corridor is not sanitizable. Please remove tape and residue for a sanitizable surface.	10/30/15		10/2/15	October

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DCM Hitchcock	118 (Main area)	Deficiency	Sanitation	Mop rack needs to be installed by the unused cagewash so that all mops can be hung up.	10/30/15		WO submitted 11/5/15	October
DCM HR&T Vivarium	Clean Washroom	Deficiency	Sanitation	Room appears to be used as a storage room. Room is cluttered with materials stored on the floor or on unmovable racks against the wall. Cages are being stacked directly on the floors and not on racks. Bedding container used to fill cages does not have a lid. The floor is dirty under racks and the walls cannot be evaluated. Please clean the floors and the walls.	8/17/15		8/17/15	July
DCM HR&T Vivarium	Feed Room	Deficiency	Sanitation	There are no records that the feed barrel has been cleaned for a long period of time. There is loose feed in the bottom of the barrel.	8/17/15		8/17/15	July
DCM Kincaid	558 (Chamber 5A)	Deficiency	Sanitation	Computer and cardboard on the floor. Please move and store up off the floor.	10/28/15		10/6/15	October
DCM K-Wing Vivarium	Clean Cage Wash	Deficiency	Sanitation	Floor is very dirty, especially the corners, but in numerous areas. Please establish a regular mopping schedule and evaluate the soap being used.	8/7/15		7/28/15	July
DCM K-Wing Vivarium	Halls	Deficiency	Sanitation	Halls are dirty along the coving. Please clean.	8/7/15		7/31/15	July
DCM K-Wing Vivarium	I012Q	Deficiency	Sanitation	Paper tag and tape on hood are not sanitizable. This was corrected during the visit.	Corrected during visit		7/8/15	July
DCM K-Wing Vivarium	I015B	Deficiency	Sanitation	Paper sign and paper backed tape on step stool needs to be removed.	8/7/15		7/22/15	July
DCM K-Wing Vivarium	I015M	Deficiency	Sanitation	Tape residue on chair needs to be removed or chair needs to be replaced.	8/7/15		8/7/15	July
DCM K-Wing Vivarium	I015M	Deficiency	Sanitation	Post-It note on hood either needs to be identified as temporary or replaced with a sanitizable solution.	8/7/15		7/29/15	July
DCM K-Wing Vivarium	I015N	Deficiency	Sanitation	Paper taped on door was removed during the visit, but tape residue will need to be removed.	8/7/15		7/22/15	July
DCM K-Wing Vivarium	K017B, Halls	Deficiency	Sanitation	Autoclave tape residue is on many containers in the halls and in K017B. Please remove tape residue.	8/7/15		8/10/15	July
DCM Roosevelt	255	Deficiency	Sanitation	Air vents on hood surface are dirty and need to be cleaned.	8/6/15		8/6/15	July
DCM Roosevelt	254, 255	Deficiency	Sanitation	Tape affixing hex wrench to the hood is not sanitizable. Please remove tape.	8/6/15		8/6/15	July

Deficiencies-at-a-Glance by Type
July - Oct 2015

DCM SLU 3.1	SB087A	Deficiency	Sanitation	Plastic debris is littered on the floor. Please dispose of debris.	8/31/15		7/22/15	July
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Sanitation	Plastic bags, debris, and folded cardboard box from behind floor chest freezer must be removed. These items provide potential harborage for vermin and are a potential fire hazard.	10/30/15		10/30/15	September
Friday Harbor Labs	Friday Harbor Labs	Deficiency	Sanitation	Rusted metal clips used to secure laminated tank labels should be replaced with double-sided Velcro strips secured to each tank.	11/13/15		11/2/15	September
Friday Harbor Labs	Lab 5	Deficiency	Sanitation	Cardboard boxes on the shelves above sink that are being used for equipment/supply storage are not sanitizable. Please remove or replace with sanitizable containers.	10/30/15		10/30/15	September
Friday Harbor Labs	Lab 6	Deficiency	Sanitation	Cardboard boxes are not sanitizable. Cardboard must be replaced with plastic containers.	10/30/15		11/6/15	September
Guthrie	351	Deficiency	Sanitation	Chairs with holes are not sanitizable. Please repair, replace, or remove the chairs.	11/10/15		11/6/15	October
Guthrie	354	Deficiency	Sanitation	Cardboard on the floor is not sanitizable. Please move off the floor or remove from the room.	11/10/15		10/27/15	October
Guthrie	355	Deficiency	Sanitation	Strip of old tape adhered to the back left wall is not sanitizable. Please remove tape and tape residue for a sanitizable surface.	11/10/15		11/6/15	October
Guthrie	052	Deficiency	Sanitation	Cardboard and pegboard are not sanitizable and should be removed from the surgery area.	11/10/15		11/6/15	October
Guthrie	351C	Deficiency	Sanitation	Brooms resting on the floor need to be stored off the ground and hung on a hook or moved out of the animal use area.	11/10/15		11/6/15	October
Guthrie	364 (Euthanasia, Dirty Cage Storage)	Deficiency	Sanitation	The mop was in the mop bucket rather than stored hanging. Please hang mops on wall mount to dry.	11/10/15		11/10/15	October
Guthrie	369B	Deficiency	Sanitation	Space is not currently housing animals, but drapes are discarded on the floor. Please keep room tidy and drapes either discarded in bins or stored.	11/10/15		11/6/15	October
HR&T	315D	Deficiency	Sanitation	Paper calendar to the left of the sink is not sanitizable and needs to be encased in plastic.	8/6/15		8/6/15	July
HR&T	B004	Deficiency	Sanitation	Cardboard boxes on shelves above surgical area need to be removed.	8/6/15		7/16/15	July

Deficiencies-at-a-Glance by Type
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HR&T	B016	Deficiency	Sanitation	Unfinished wood board was removed during the visit.	Corrected during visit		7/15/15	July
HR&T	B016	Deficiency	Sanitation	Cardboard boxes on shelves need to be removed.	8/6/15		8/6/15	July
HSB	B108B	Deficiency	Sanitation	Metal cart used to transport animals is rusty and dirty. The cart needs to be cleaned prior to use.	Prior to use		10/2/15	September
HSB	BB1553	Deficiency	Sanitation	Two chairs with non-sanitizable backsides. Please remove and replace with chairs that have sanitizable surfaces.	10/28/15		10/23/15	October
HSB	BB1553, BB1604	Deficiency	Sanitation	Paper posted on the door in BB1553 and on wall (map, eyewash log) in BB1604 are not sanitizable. Paper must be laminated or encased in plastic.	10/28/15		10/23/15	October
HSB	BB1577	Deficiency	Sanitation	Cardboard boxes storing lab supplies are under the counter opposite to animal work area are not sanitizable. The cardboard should be removed or be replaced with plastic storage containers.	11/9/15		10/27/15	October
HSB	BB857D	Deficiency	Sanitation	Two pieces of paper are not encased in plastic. This was corrected during the visit.	Corrected during visit		10/1/15	October
HSB	D218	Deficiency	Sanitation	Cardboard box with paper, binder clips, and duct tape are being used to perform a videotaped locomotion test with some of the mice prior to euthanasia. The cardboard, tape and paper are not sanitizable, and the box has an open end that allows rodents to potentially escape in the lab. The cardboard box should be replaced with a plastic tub that is sanitizable and safe for the animals.	11/5/15		11/4/15	October
HSB	E064B	Deficiency	Sanitation	Floor is very dusty/dirty and should be cleaned. Please develop a cleaning schedule.	11/6/15		11/4/15	October
HSB	E064B	Deficiency	Sanitation	Metal stool in room is very rusty and not sanitizable. Please remove or replace stool.	11/6/15		11/4/15	October
HSB	H507	Deficiency	Sanitation	Floor is dirty. Some hazards are on the floor (e.g., glass coverslips) as well as other lab supplies.	11/6/15		11/4/15	October
HSB	I201B	Deficiency	Sanitation	Mops and brooms used for cleaning must be stored off the ground and hung on a hook or moved out of the animal use area.	11/10/15		11/11/15	October

Deficiencies-at-a-Glance by Type
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HSB	I201B	Deficiency	Sanitation	Cardboard boxes stored on the floor are not sanitizable. Cardboard must be removed or replaced with plastic containers.	11/10/15		10/20/2015	October
HSB	I201B	Deficiency	Sanitation	Floor is dirty. Please clean and develop a cleaning schedule.	11/10/15		10/21/15	October
HSB	I201B	Deficiency	Sanitation	Room is cluttered and should be kept clean. Tape residue is not sanitizable and needs to be removed.	11/10/15		10/20/15	October
HSB	K227, K233	Deficiency	Sanitation	Eyewash log needs to be encased in a sanitizable plastic sleeve.	10/14/15		10/8/15	September
HSB	RR419	Deficiency	Sanitation	Paper tape is not sanitizable. Please remove and replace with a sanitizable alternative.	11/9/15		11/9/15	September
HSB	RR813	Deficiency	Sanitation	Paper taped on wall. Tape is not sanitizable and needs to be removed. Paper needs to be laminated or encased in plastic.	10/12/15		10/12/15	September
HSB	RR813	Deficiency	Sanitation	Cardboard on the floor is not sanitizable. Please move off the floor or remove from the room.	10/12/15		10/12/15	September
HSB	T163N	Deficiency	Sanitation	Paper posted on fume hood is not sanitizable. Paper must be laminated or encased in plastic. Tape can be used if changed frequently and there is no tape residue remaining.	8/12/15		8/12/15	July
Kincaid	Roof Aviaries	Deficiency	Sanitation	Several locations on the roof away from the aviaries have piles of trash/discarded items/old aviary components. Please haul away trash and keep the area clutter-free.	11/6/15		11/16/15	October
Manchester	Halibut Pad	Deficiency	Sanitation	Disintegrating sun shades/covers need to be replaced prior to use.	Prior to use		9/24/15	August
SLU	N101	Deficiency	Sanitation	Induction box appears to have the edges reinforced with glue, however, the nozzle is loose and is attached with tape, which is not sanitizable. This poses an occupational hazard. Recommended the group to purchase a new induction box.	9/30/15		9/21/15	August
SLU	N101, N115	Deficiency	Sanitation	Brooms resting on the floor need to be stored off the ground and hung on a hook or moved out of the animal use area.	9/4/15		9/4/15	August
SLU	S462	Deficiency	Sanitation	Tape on the surgical table is not sanitizable. Please remove tape for a sanitizable surface.	8/28/15		8/27/15	August

Deficiencies-at-a-Glance by Type
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SLU	S462	Deficiency	Sanitation	Animal droppings found on floor, and fur in corners and along the sides of imaging chambers. There is no SOP or cleaning log in place. Please develop an SOP and plan for cleaning.	8/28/15		8/27/15	August
SLU	S485	Deficiency	Sanitation	Cardboard encased containers on the floor by the door are not sanitizable. The cardboard boxes must be raised up off the floor or removed the room.	9/10/15		9/1/15	August
SLU	S546	Deficiency	Sanitation	Microscope curtain has tears in the corners and is unsanitizable. Please replace prior to use.	Prior to use		11/17/15	September
SLU	S546	Deficiency	Sanitation	Unsealed paper items present. This was corrected during the visit.	Corrected during visit		9/3/15	September
SLU	S573	Deficiency	Sanitation	Old tape labels on shelves are not sanitizable. Please replace with a sanitizable alternative (e.g., laminated labels held with magnets).	8/28/15		8/26/15	August
SLU 3.1	E258	Deficiency	Sanitation	Eyewash log needs to be encased in a sanitizable plastic sleeve.	8/21/15		8/25/15	August
WaNPRC HSB Lab	I013	Deficiency	Sanitation	Cardboard and foam padding stored near terminal surgery is non-sanitizable and must be removed.	10/12/15		10/6/15	September
WaNPRC I-Wing	I040M, Corridor	Deficiency	Sanitation	Duct tape on the smoke detector needs to be removed. This was corrected during the visit.	Corrected during visit		9/9/15	September
WaNPRC I-Wing	I059	Deficiency	Sanitation	Tape residue on the outside of the door needs to be removed. Residue is not sanitizable.	10/21/15		10/13/15	September
WaNPRC I-Wing	I455	Deficiency	Sanitation	Chair with holes in it is not sanitizable. Please repair or remove the chair.	10/21/15		10/19/15	September
WaNPRC I-Wing	I455A	Deficiency	Sanitation	Paper on the cupboard is not sanitizable and needs to be covered with plastic. This was corrected during the visit.	Corrected during visit		9/9/15	September
WaNPRC I-Wing	I565, I567, I733, I737	Deficiency	Sanitation	Procedure request form is not sanitizable. Paper must be encased in plastic sleeves.	10/20/15		10/20/15	September
WaNPRC RR-Wing	Feed and Bedding Room	Deficiency	Sanitation	Two racks had rusty casters. Please remove the rust or remove racks.	10/26/15		11/3/15	September

Deficiencies-at-a-Glance by Type
July - Oct 2015

WaNPRC RR-Wing	Feed and Bedding Room	Deficiency	Sanitation	Food and bedding room dirty in corners. There is difficulty assessing and accessing corners as room is very crowded with non-food items. Cannot reach the back of the room due to too many racks being present. Cannot assess vermin well in back of room as cannot see floor.	11/9/15		11/9/15	September
WaNPRC RR-Wing	RR148	Deficiency	Sanitation	Floor under the sink is dirty. Please develop a cleaning schedule.	10/19/15		9/21/15	September
WaNPRC Western	239	Deficiency	Sanitation	Tape residue on storage cart is not sanitizable. Please remove.	11/10/15		10/27/15	September
WaNPRC Western	320	Deficiency	Sanitation	Paper on wall is not sanitizable. Paper must be laminated or encased in plastic.	11/10/15		10/27/15	September
WaNPRC Western	327	Deficiency	Sanitation	Room is cluttered. Please try to make the room less crowded to allow more space for work.	10/27/15		10/27/15	September
WaNPRC Western	327	Deficiency	Sanitation	Notebooks and paper need to be stored separate from animals.	11/10/15		10/27/15	September
WaNPRC Western	Necropsy Room	Deficiency	Sanitation	Large puddle of water on the floor. A mechanism for cleaning up needs to be in place. Brooms and mop need to be hung up off the floor.	10/27/15		10/27/15	September
WaNPRC Western	Throughout facility	Deficiency	Sanitation	Cleaning implements need to be stored off of the floor. Hangers need to be supplied where needed.	10/27/15		10/27/15	September
WaNPRC Western	WaNPRC Western	Deficiency	Sanitation	Mops and sponges need to be replaced. Several are soiled and/or worn out. Recommend regular replacement schedule rather than relying on an as needed basis.	10/27/15		10/27/15	September

**Analysis of deficiencies found
University of Washington IACUC Site Visits July-October 2015**

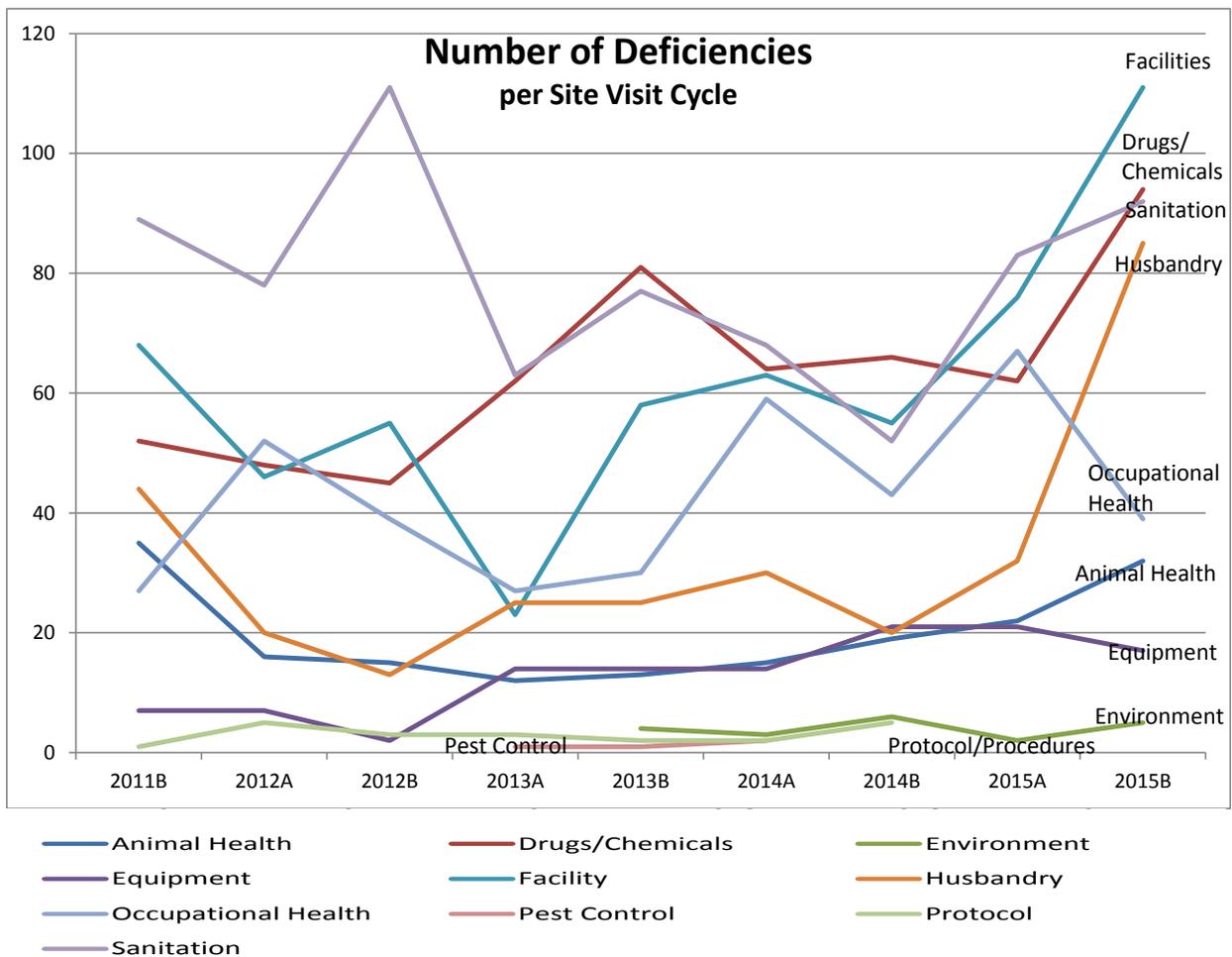
Summary, second half 2015

Degree	PI Labs	DCM	WaNPRC	Grand Total
Significant	1			1
Deficiency	212	174	89	475
Grand Total	213	174	89	476

Significant Deficiencies, second half 2015

Degree	Type	Deficiency
Significant	Protocol/Procedure	Post-surgical analgesia is not being administered according to approved protocol. Time between doses is too short or too long, and not being administered for the full 48 hours. Scientific Liaison will contact group regarding potential for adding SR buprenorphine to protocol.

Running tallies for the past four years



Deficiencies 2015 (first half, second half)

type	PI Labs		DCM		WaNPRC		total	
	2015A	2015B	2015A	2015B	2015A	2015B	2015A	2015B
Animal Health	7	5	13	20	2	7	22	32
Drugs / Chemicals	13	48	39	34	10	12	62	94
Environment		2	1		1	3	2	5
Equipment	4	7	16	7	1	3	21	17
Facility	13	48	38	26	25	37	76	111
Husbandry	4	29	26	49	2	7	32	85
Occupational Health	17	27	46	8	4	4	67	39
Protocol		1					0	1
Sanitation	27	46	46	30	10	16	83	92
Total	85	213	225	174	55	89	365	476

Discussion of the top categories

The latest site visit cycle saw a significant increase in the total number of deficiencies found. This may be attributable to both increased training of the IACUC and to increased focus in advance of our triennial AAALAC accreditation visit, which occurred the third week of November. All categories of deficiency saw increases, with the notable exception of Occupational Health. Also, a large increase in Husbandry deficiencies elevated this category into the top four, alongside Facilities, Drugs/Chemicals, and Sanitation.

Facilities

Walls, doors, ceilings and floors that need repair accounted for 55, or almost half, of the Facility deficiencies. Fixtures (built-in cabinets, counters, racks, etc.) needing repair or having unsanitizable surfaces (rusty, scratched, etc.) account for an additional 37%. Other deficiencies included issues related to plumbing, electrical and ventilation. Both PI Labs and WaNPRC saw increases in Facility deficiencies from the first half of 2015, while DCM saw a decrease – from 38 to 27 citations.

Drug and Chemical Storage and Documentation

The Drugs/Chemicals category was cited for 95 deficiencies in the second half of 2015. Expired drugs are still the top problem (41% of the category), followed by incomplete or missing labeling of drugs (17%). Problems with cleaning supplies and other chemicals accounted for 18% of deficiencies in this category, with similar predominant causes -- expired products or incomplete or missing labeling. PI Labs accounted for 48 of the deficiencies in this category, followed by DCM spaces with 35, and WaNPRC with 12.

Sanitation

Sanitation deficiencies were cited 90 times. The largest number, 27 or 30%, related to an ongoing bugaboo: taped up paper signs that cannot be sanitized. The next largest issue, 23 citations, was floors, counters and other surfaces in need of cleaning. Non-sanitizable equipment

and furnishings were noted 14 times, and another old bugaboo, improperly used cardboard containers, were cited 13 times. Notably, no cardboard containers were cited in DCM spaces!

Husbandry

The large jump in Husbandry deficiencies stemmed from numerous causes. Tasks not being logged properly and incomplete or missing cage cards both increased significantly, and respectively accounted for 27% and 19% of all citations in this category. Missing items -- such as environmental enrichment, required signage, animal care variances, and cage drapes -- also increased and accounted for 16% of the category. Other Husbandry deficiencies included not adhering to cleaning schedules, crowded cages, and unreported temperature/humidity fluctuations. Husbandry deficiencies occurred across DCM, PI and WaNPRC spaces.

Occupational Health

The decrease in Occupational Health issues is due to a notable reduction of deficiencies within DCM spaces. For DCM, unflushed eyewash stations, recapped needles, overfilled sharps containers unsecured gas cylinders, and expired materials were greatly reduced or non-existent in the last half of 2015. Kudos!

Appendix C – Multiple Major Surgeries

Protocol Number	Multiple Major Surgeries
2048-02	<p>Multiple Major Surgery: 1. Electrophysiological recording and injection of plasmids or AAVs 2. Either dorsal cochlear tract cut or cochlear ablation, 2-4 days following #1 Species: Chickens Interval: 1 hour-4 days Rationale: Surgery 1 is needed to treat with the plasmids to label the proteins of interest. Surgery 2 is needed to do the manipulation (e.g., dorsal tract cut) to observe the effect on the labeled proteins.</p> <p>OR</p> <p>Multiple Major Surgery: 1. Craniotomy to inject TTX 2. Craniotomy to inject TTX, 4 hours after #1 3. Terminal cochlear ablation and ABR test before perfusion, 4 hours to 7 days after #2 Species: Chickens Interval: 4 hours to 7 days Rationale: Surgeries 1 and 2 are needed to treat with TTX (8 hours of blockade is required, and a single TTX injection lasts 4 hours). Surgery 3 is the test for function following the prolonged blockade. Maintaining anesthesia for 12 hours would be too risky, and a recovery is required for intervals up to 7 days.</p> <p>Permissible Weight Loss Variance: Species: Mice and Rats Rationale: We are approved to administer aminoglycoside antibiotics to rats and mice. AG administration often results in a temporary drop in weight. Animals typically recover their weight a week or two following treatment. During treatment, animals are weighed and monitored daily. If weight loss is observed, animals are given nutritive supplements and saline injections to assist their recovery. We wish to adjust our current euthanasia criteria to allow for veterinary consultation when an animal's weight loss approaches 20%. In this way, if an animal is showing signs of improvement and meets with veterinary approval, supportive measures may be continued and the animal may remain in the study. If rats reach 20% weight loss, Veterinary Services will be contacted to determine if additional supportive care is appropriate or euthanasia is warranted on a case-by-case basis. Between 20-30% weight loss, supportive care and monitoring will be done in conjunction with Vet Services, with ongoing assessment to determine if the rat is at a humane endpoint. Rats will be euthanized if they reach 30% weight loss despite intensive supportive care.</p>
2161-24	<p>Multiple Major Surgery: 1. Electrode implantation for EEG (epidural or depth), may include other minor surgeries such as ECG, EMG. 2. Electrode implantation for EEG (epidural or depth), more than 48 hrs following #1 Species: Mice Interval: > 48 hrs Rationale: Individual animals will be followed over an extended period of time collecting several EEG/video recordings in order to test the effects of special diet, anti-epileptic drugs and/or thermal induction over time. This may require 1 repair surgery so that valuable data is not lost due to early termination of the experiment resulting from the electrode being dislodged.</p> <p>OR</p> <p>1. Implant of E-mitter 2. Repair and/or replacement of E-mitter, more than 48 hrs following #1 Species: Mice Interval: > 48 hrs Rationale: Individual animals will be followed over an extended period of time collecting simultaneous activity, temperature, heart rate, and video data in order to evaluate whether hyperthermia induced seizures</p>

Appendix C – Multiple Major Surgeries

	<p>and spontaneous seizures occur at the same temperature, and how seizures/ antiepileptic drugs affect normal sleep behaviors. This may require 1 repair surgery so that valuable data is not lost due to early termination of the experiment resulting from the E-mitter being dislodged.</p>
2183-02	<p>Multiple Major Surgery: 1. Intracranial Virus Injection 2. Intracranial Virus Injection, at least one week after #1 Species: Mice Interval: At least one week Rationale: We often use viral vectors to insert genetic components that interact with one another. For example, we can activate one population while inhibiting a second population. Since we produce all our viral vectors in-house this would save on a limited resource to be able to first make sure the initial viral injection was successfully targeted by performing an already approved behavioral test (often we look for an increase or decrease in feeding) before going ahead and injecting a second virus to manipulate a second population of neurons.</p> <p>OR</p> <p>Multiple Major Surgery: 1. Intracranial Virus Injection 2. Brain Cannulation, at least one week after #1 Species: Mice Interval: At least one week Rationale: We implant Fiber optic cannulae in order to shine light onto neurons and either activate or inhibit specific neuronal populations or targets using virally delivered light-activated ion channels. We have developed a strategy that would allow us to behaviorally prescreen animals that had successful viral targeting and allow ample time for viral expression (2-3weeks) before implanting fiber optic cannulae. These simple tests take less than one week and use our BioDaq cages which are incompatible with the animals having a cannula implant attached to their skull. This will cut down on the time the animals will spend with the implanted cannula affixed to the skull. This would be a great benefit, because 1) over time scar tissue can block the efficiency of the light to reach neurons and 2) with more time, mice are more likely to knock the implants loose or damage the connection needed to couple to the fiber optic laser.</p>
2195-27	<p>Multiple Major Surgery: 1. Laparotomy 2. Laparotomy, approximately 1 month, with a minimum of at least 3 weeks following #1 3. Laparotomy, approximately 1 month, with a minimum of at least 3 weeks following #2 Species: M. Nemestrina Interval: Approximately 1 month, with a minimum of at least 3 weeks Rationale: We will assess the progression of chlamydial pathology in the upper reproductive tract (URT) after each macaque has undergone direct tubal and endocervical inoculations with <i>C. trachomatis</i>.</p>
2224-05	<p>Multiple Major Surgery: 1. Total right nephrectomy 2. Partial resection of left kidney via bilateral resection of poles, 1 week following #1 Species: Mice Interval: 1 week Rationale: It is necessary to perform a 2-step renal surgery model because the animals will not recover if both surgeries are done at the same time. Two surgeries provides for better survival and better induction of uremia.</p>
2225-04	<p>Multiple Major Surgery: 1. Thoracotomy and myocardial infarction 2. Thoracotomy with intervention and placement of telemetry device (rats), 4 days to 2 weeks following #1 Species: Mice and Rats Interval: 4 days to 2 weeks Rationale: These studies involve one surgery to induce myocardial infarction and a second surgery to either inject a cell suspension or to place engineered myocardium or a scaffold into the injury during the healing phase. In some studies we are investigating the electrical integration of the implanted cells with the host heart.</p>

Appendix C – Multiple Major Surgeries

	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Thoracotomy and myocardial infarction 2. Thoracotomy with intervention, 1 week following #1 <p>Species: Rabbits Interval: 1 week</p> <p>Rationale: In many cases it will be necessary to perform two survival surgeries in one animal. These studies involve one surgery to induce myocardial infarction and the second to inject cells into the injury during the healing phase. Rabbits are not large enough to administer cells through an interventional catheter (directly into the injured heart) and injecting cells at a later time point is a relevant model for future clinical studies.</p>
2225-06	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Myocardial infarction surgery and Vmed electrode and wire implantation 2. Cell therapy injection (by thoracotomy or catheter based approach), cardiac monitor devise (telemetry) insertion, and possible lead placement in the thorax 12-16 days following #1 3. Thoracotomy, atrial scar creation, electrode implants, 28 days following #2 (this applies to 2 animals only) <p>Species: Macaques Interval: 12-16 days</p> <p>Rationale: These studies are designed to investigate the therapeutic effect, the optimal dose and cell-delivery route, and the mechanism of ventricular arrhythmias after cell therapy in myocardial infarction animals. It is necessary to create the myocardial infarction before delivering the therapy. The infarcted animal's cardiac function will stabilize 2 weeks after myocardial infarction. We will do cell therapy approximately 2 weeks after myocardial infarction surgery. In our previous studies we have had animals showing ventricular arrhythmias between 1 week and 4 weeks after intramyocardial cell injection. Because ventricular tachycardia may cause death in humans, it is necessary to figure out the etiologies of hESC-CM transplant-related ventricular arrhythmia and solve this problem. The terminal thoracotomy has been added to test the feasibility of administering stem cells 2-4 weeks after the initial injury for future studies.</p>
2326-08	<p>Multiple Major Survival Surgery:</p> <p>Path A</p> <p>Pre-surgical procedure: MRI (anesthesia only)</p> <ol style="list-style-type: none"> 1. Skull Surgery: HS 2. Skull Surgery: CI, minimum 1 week after #1 3. Arm EMG Implant I, minimum 1 week after #2 4. Subcutaneous (SubQ) wires from head-back I, minimum 1 week after #3 5. Repair Surgery I, amount of time will depend on need, but will be preceded by veterinary and pre-survival assessment <p>Interval: minimum of 1 week</p> <p>Rationale for the above surgeries in a single animal: Where possible, multiple procedures are performed during a single surgery.</p> <p>For some animals, the BCI implant is custom-machined to a physical mold of each monkey's skull, to optimize fit and minimize weight and size. This machining process necessitates a delay between surgeries 1 and 2. In some animals we will also use this period to map the physiological properties of a brain area to aid placement of permanent electrodes in surgery 2.</p> <p>Surgeries 3 and 4, if performed, require separate surgeries due to their length and complexity. Veterinarians at the Primate Center concur with the use of multiple surgeries to implant these devices. These surgeries involve exposure of separate areas, so no time would be saved by combining either with any other procedure. Such a combined surgery would require an excessive period of continuous anesthesia that could adversely affect cardiac or respiratory function, increase risk of mortality during surgery, and prolong post-surgical recovery.</p> <p>Subcutaneous wires (surgery 4) are needed only if transcutaneous EMGs are used, in order to connect to the implant in Skull Surgery: CI, to monitor EMG activity during free behavior.</p> <p>Specific experiments that will use this surgical chain This path concerns experiments that involve cortical recording or stimulation, in the booth or in free behavior, sometimes in conjunction with muscle (EMG) recording.</p>

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	<ol style="list-style-type: none"> 1. Operant conditioning of cortical activity, in the booth or in free behavior (Experiment group A-I) 2. Cortical activity-triggered cortical stimulation, in the booth or in free behavior (Experiment group A-I) 3. Paired-pulse cortical stimulation, in the booth or in free behavior (Group A-I) 4. EMG-triggered cortical stimulation, in the booth (Experiment group A-II) 5. EMG-triggered cortical stimulation in free behavior (Experiment group A-III) <p>Path B.</p> <ol style="list-style-type: none"> 1. Skull Surgery: HS (Halo Straps only) 2. Skull Surgery: CI, minimum 4 weeks after #1 3. Arm EMG implant, minimum 1 week after #2 4. Subcutaneous (SubQ) wires from head-back, minimum 1 week after #3 5. Spinal implant, minimum 1 week after #4 6. Repair surgery, amount of time will depend on need, but will be preceded by veterinary and pre-survival assessment <p>Interval: minimum 1-4 weeks</p> <p>Rationale for the above surgeries in a single animal: Where possible, multiple procedures are performed during a single surgery.</p> <p>For some animals, the BCI implant is custom-machined to a physical mold of each monkey's skull, to optimize fit and minimize weight and size. This machining process necessitates a delay between surgeries 1 and 2. In some animals we will also use this period to map the physiological properties of a brain area to aid placement of permanent electrodes in surgery 2.</p> <p>Surgeries 3, 4 and 5, if performed, require separate surgeries due to their length and complexity. Veterinarians at the Primate Center concur with the use of multiple surgeries to implant these devices. These surgeries involve exposure of separate areas, so no time would be saved by combining either with any other procedure. Such a combined surgery would require an excessive period of continuous anesthesia that could adversely affect cardiac or respiratory function, increase risk of mortality during surgery, and prolong post-surgical recovery.</p> <p>Subcutaneous wires (surgery 4) are needed only if transcutaneous EMGs are used, in order to connect to the implant in Skull Surgery: CI, to monitor EMG activity during free behavior.</p> <p>Specific experiments that will use this surgical chain This path includes experiments that may involve spinal recording and/or stimulation in the booth or in free behavior, sometimes in conjunction with cortical recording and/or stimulation or muscle (EMG) recording.</p> <ol style="list-style-type: none"> 1. Operant conditioning of cortical activity, in the booth or in free behavior (Experiment group B-I) 2. Cortical activity-triggered cortical stimulation, in the booth or in free behavior (Experiment group B-I) 3. Paired-pulse cortical stimulation, in the booth or in free behavior (Group B-I) 4. EMG-triggered cortical stimulation, in the booth (Experiment group B-II) 5. EMG-triggered cortical stimulation in free behavior (Experiment group B-III) 6. Cortical activity-triggered spinal stimulation, in the booth or in free behavior (Experiment group B-IV) 7. Spinal activity-triggered spinal stimulation, in the booth or in free behavior (Experiment group B-IV) 8. EMG activity-triggered spinal stimulation, in the booth or in free behavior (Experiment group B-IV) 9. Paired-pulse cortical and spinal stimulation, in the booth or in free behavior (Experiment group B-IV) 10. Measurement of spinal stimulation output via stimulus-evoked EMG activity, in the booth or in free behavior (Experiment group B-IV) <p>Path C</p> <ol style="list-style-type: none"> 1. Skull Surgery: HS 2. VNS implant, minimum 4 weeks after #1 3. Arm EMG implant, minimum 1 week after #2
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	<p>4. Spinal implant, minimum 1 week after #3</p> <p>5. Repair Surgery, amount of time will depend on need, but will be preceded by veterinary and pre-survival assessment</p> <p>Interval: minimum 1-4 weeks</p> <p>Rationale for the above surgeries in a single animal: Where possible, multiple procedures are performed during a single surgery.</p> <p>Surgeries 2, 3 and 4, if performed, require separate surgeries due to their length and complexity. Even though surgery 4 is relatively short, surgeries 3 and 5 are particularly lengthy and complex, making it impossible to perform in combination. Veterinarians at the Primate Center concur with the use of multiple surgeries to implant these devices. These surgeries involve exposure of separate areas, so no time would be saved by combining either with any other procedure. Such a combined surgery would require an excessive period of continuous anesthesia that could adversely affect cardiac or respiratory function, increase risk of mortality during surgery, and prolong post-surgical recovery.</p> <p>Subcutaneous wires (surgery 4) are needed only if transcutaneous EMGs are used, in order to connect to the implant in Skull Surgery: CI, to monitor EMG activity during free behavior.</p> <p>Specific experiments that will use this surgical chain This path concerns experiments in the booth, that involve cortical recording and/or stimulation using a cortical chamber (see description of Surgery HS), sometimes in conjunction with muscle (EMG) recording or spinal recording and/or stimulation, and VNS.</p> <ol style="list-style-type: none"> 1. Cortical activity-triggered cortical stimulation (Experiment group C-I) 2. Cortical activity-triggered cortical stimulation, with and without VNS (within subject comparison), in booth (Experiment group C-II) 3. Paired-pulse cortical stimulation, with and without VNS (within subject comparison), in booth (Experiment group C-II) 4. EMG-triggered cortical stimulation, with and without VNS (within subject comparison), in booth (Experiment group C-III) 5. Cortical activity-triggered spinal stimulation, in the booth (Experiment group C-IV) 6. Spinal activity-triggered spinal stimulation, in the booth (Experiment group C-IV) 7. EMG activity-triggered spinal stimulation, in the booth (Experiment group C-IV) 8. Paired-pulse cortical and spinal stimulation, in the booth (Experiment group C-IV) 9. Measurement of spinal stimulation output via stimulus-evoked EMG activity, in the booth (Experiment group C-IV) 10. Cortical activity-triggered spinal stimulation, with and without VNS (within subject comparison), in booth (Experiment group C-IV) 11. Paired-pulse cortical and spinal stimulation, with and without VNS (within subject comparison), in booth (Experiment group C-IV) <p>Path D</p> <ol style="list-style-type: none"> 1. Skull Surgery: HS (Halo Straps only) 2. Skull Surgery: CI, minimum 4 weeks after #1 3. VNS implant, minimum 1 week after #2 4. Arm EMG implant, minimum 1 week after #3 5. Subcutaneous (SubQ) wires from head-back, minimum 1 week after #4 6. Spinal implant, minimum 1 week after #5 7. Repair surgery, amount of time will depend on need, but will be preceded by veterinary and pre-survival assessment <p>Interval: minimum 1- 4 weeks</p> <p>Rationale for the above surgeries in a single animal: Where possible, multiple procedures are performed during a single surgery.</p> <p>For some animals, the BCI implant is custom-machined to a physical mold of each monkey's skull, to optimize fit and minimize weight and size. This machining process necessitates a delay between surgeries 1 and 2. In some animals we will also use this period to map the physiological properties of a brain area to aid placement of permanent electrodes in surgery 2.</p> <p>Surgeries 3, 4, 5 and 6 if performed, require separate surgeries due to their length and complexity. Even though surgeries 3 and 5 are relatively short, surgeries 4 and 6 are particularly lengthy and complex, making it impossible to perform in combination. Veterinarians at the Primate Center concur with the use</p>
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	<p>of multiple surgeries to implant these devices. These surgeries involve exposure of separate areas, so no time would be saved by combining either with any other procedure. Such a combined surgery would require an excessive period of continuous anesthesia that could adversely affect cardiac or respiratory function, increase risk of mortality during surgery, and prolong post-surgical recovery.</p> <p>Subcutaneous wires (surgery 4) are needed only if transcutaneous EMGs are used, in order to connect to the implant in Skull Surgery: CI, to monitor EMG activity during free behavior.</p> <p>Specific experiments that will use this surgical chain This path concerns experiments that involve cortical recording and/or stimulation, in the booth or in free behavior, in conjunction with VNS or intracortical delivery of chemical substances, sometimes combined with spinal recording and/or stimulation and muscle (EMG) recording.</p> <ol style="list-style-type: none"> 1. Cortical activity-triggered cortical stimulation, with and without intracortical delivery of chemical substances (within subject comparison), in booth and in free behavior (Experiment group D-I) 2. Paired-pulse cortical stimulation, with and without intracortical delivery of chemical substances (within subject comparison), in booth and in free behavior (Experiment group D-I) 3. Cortical activity-triggered cortical stimulation, with and without VNS (within subject comparison), in booth and in free behavior (Experiment group D-II) 4. Paired-pulse cortical stimulation, with and without VNS (within subject comparison), in booth and in free behavior (Experiment group D-II) 5. EMG-triggered cortical stimulation, with and without VNS (within subject comparison), in booth (Experiment group D-III) 6. EMG-triggered cortical stimulation, with and without intracortical delivery of chemical substances (within subject comparison), in booth and in free behavior (Experiment group D-III) 7. EMG-triggered cortical stimulation, with and without VNS (within subject comparison), in booth and in free behavior (Experiment group D-IV) 8. Cortical activity-triggered spinal stimulation, with and without VNS (within subject comparison), in booth and in free behavior (Experiment group D-V) 9. Paired-pulse cortical and spinal stimulation, with and without VNS (within subject comparison), in booth and in free behavior (Experiment group D-V) 10. Cortical activity-triggered spinal stimulation, with and without intracortical delivery of chemical substances (within subject comparison), in booth and in free behavior (Experiment group D-V) <p>Path E</p> <ol style="list-style-type: none"> 1. Skull Surgery: HS (Halo Straps only) 2. Skull Surgery: CI: CI, minimum 4 weeks after #1 3. Nerve Block Implant, minimum 1 week after #2 4. Skull Surgery: CI (repeated for multiple Utah arrays, if needed) , minimum 1 week after #3 5. Nerve Block Implant, minimum 1 week after #4 6. Repair surgery, amount of time will depend on need, but will be preceded by veterinary and pre-survival assessment <p>Interval: minimum 1-4 weeks</p> <p>Rationale for the above surgeries in a single animal: Where possible, multiple procedures are performed during a single surgery (i.e.: the halo frame is attached during Skull Surgery: CI).</p> <p>For animals utilizing the halo head restraint, a delay of 4 weeks or more is ideal for surgeries 1 and 2 to allow titanium plates to osseointegrate with the animal's skull, providing increased implant stability.</p> <p>Repeating Skull Surgery: CI will allow us to compare multiple brain areas in the same animal. This will reduce the number of animals needed for the study and permit quantitative comparisons between brain areas while removing variations in skill among animals.</p> <p>Nerve Block Implants are short-lived and may need to be re-implanted up to 3 times, allowing ample recovery time between each (typically 4 weeks or more).</p> <p>Specific experiments that will use this surgical chain This path concerns experiments that involve cortical recording in conjunction with a peripheral nerve block, sometimes in conjunction with muscle stimulation.</p> <ol style="list-style-type: none"> 1. Operant conditioning of cortical activity, with and without nerve block (within animal comparison), in booth and in free behavior (Experiment groups E-I and E-III) 2. Cortical activity-triggered muscle stimulation after nerve block, in booth and in free behavior
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	(Experiment groups E-II, E-III and E-IV)
2340-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Implant eye coil and head stabilizing fitting 2. Implant recording chamber, minimum of 30 days following #1 3. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Interval: 30 days minimum Species: M. mulatta</p> <p>Rationale: Surgery 1 is necessary to train the monkey to make the eye movements for this study and, later, to collect the movement data for this study. Surgery 2 is necessary to record the activity of brain neurons. We need to do this to locate specific brain structures and to collect the neuron response data for this study. Surgery 3 Consistent with the Guidelines for Non-human Primate Neuroscience Studies in the policies of the UW Office of Animal Welfare I request 1 provisional repair surgery for each animal new to the protocol. We could use this surgery to repair eye coils, stabilizing hardware, or a recording chamber. This request is for new monkeys only. Carry-over monkeys will come with their own repair surgeries previously approved. Interval depends on need, but is always preceded by a veterinary pre-surgical assessment.</p> <p>Multiple Major Surgery:</p> <p>NOTE: A09044: The USDA multiple surgery exemption has expired for this monkey – no more surgeries can be performed.</p> <ol style="list-style-type: none"> 1. Implant eye coil and head stabilizing fitting 2. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Interval: 30 days minimum Species: M. mulatta</p> <p>Rationale: Surgery 1 is necessary to train the monkey to make the eye movements for this study and, later, to collect the movement data for this study. Surgery 2 Consistent with the Guidelines for Non-human Primate Neuroscience Studies in the policies of the UW Office of Animal Welfare I request 1 provisional repair surgery for each animal new to the protocol. We could use this surgery to repair eye coils, stabilizing hardware, or a recording chamber (for animals in the cerebellar study). This request is for new monkeys only. Carry-over monkeys will come with their own repair surgeries previously approved. Interval depends on need, but is always preceded by a veterinary pre-surgical assessment.</p>
2348-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Thymectomy 2. Thymus grafting <p>Species: Mice Interval: 3 weeks</p> <p>Rationale: Removal of thymus to ascertain function of implanted thymic lobes. The time between the surgeries is required for the maturation of newly emigrated T cells, so the thymectomy and thymus grafting cannot be performed at the same time.</p>
2430-02	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Lesion of optic tract or thalamus or enucleation 2. Craniotomy to inject anatomical tracers, minimum of 2 weeks following #1 <p>Species: Rats Interval: 2 weeks minimum</p> <p>Rationale: The second surgery is necessary for administering the anatomical tracers that will reveal the effect that early deprivation of retinal input has on the development of the brain.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Craniotomy to inject anatomical tracers to reveal callosal pattern in opposite hemisphere 2. Craniotomy to implant electrodes for electrophysiological recording and injection of different tracer in the same recording session, minimum of 2-3 days following #1

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	<p>Species: Rats Interval: 2-3 days Rationale: Anatomical features revealed with the first tracer injections will be correlated with electrophysiological data. The first tracer injections are not done during the recording session because in preliminary experiments, the group discovered that the recording procedure reduces the transport of the tracer used. During the recording session, some sites will be identified electrophysiologically and injected with specific tracers. Survival time from the recording session is required to allow transport time for the tracers injected during recording. The second tracer injection will provide data about connectivity of identified sites.</p>
<p>2456-06</p>	<p>Multiple Major Surgery: 1. Craniotomy (2 bur-holes) for delivery of AAV-DIO-TVA-RG 2. Craniotomy (bur-hole) for delivery of rabies viral vector (SAD?G-EGFP (EnvA), a minimum of 21 days following #1 Species: Mice Interval: 21 days minimum Rational: To allow sufficient time for TVA-RG to infect and express in neurons.</p> <p>Multiple Major Surgery: 1. Hepatic branch vagotomy 2. Intra-parenchymal injections/Brain cannulation and catheterization of carotid artery and jugular vein, a minimum of 7 days following #1 Species: Rats Interval: 7 days minimum Rationale: This laboratory focuses on central mechanisms regulating energy balance and insulin sensitivity. In this study, the aim is to determine whether the hypothalamic effect of leptin on peripheral insulin sensitivity involves activation of hepatic efferent vagal fibers. To accomplish this, the lab will perform selective hepatic branch vagotomy (HV) or a sham operation in leptin receptor-deficient (Koletsy) rats. A second surgical session is needed in which animals will receive intra-parenchymal injections and be implanted with a catheter to the right jugular vein and left carotid artery. Seven days later, euglycemic-hyperinsulinemic clamp studies will then be performed to measure sensitivity. The reason for this second surgical session is that the hepatic vagotomy is expected to have small but significant changes on food intake and body weight that will significantly affect insulin sensitivity. Therefore it is necessary to give sufficient time for animals to recover before performing intra-parenchymal injections and implanting catheters. If all surgeries were conducted in one single session, if changes in insulin sensitivity using the clamp were detected, the interpretation of the experiments would be impossible because of the inability to distinguish whether these effects were due to the experimental treatment or because animals simply lost weight after surgery.</p> <p>OR</p> <p>1. Subdiaphragmatic vagal deafferentation 2. Intra-parenchymal injections/Brain cannulation and catheterization of carotid artery and jugular vein, a minimum of 7 days following #1 Species: Rats Interval: 7 days minimum Rationale: This laboratory focuses on central mechanisms regulating energy balance and insulin sensitivity. In this study, the aim is to determine whether afferent vagal fibers are required for the effects of hypothalamic effect of leptin on peripheral insulin sensitivity. To accomplish this, the lab will to perform selective vagal deafferentation or a sham operation in leptin receptor-deficient (Koletsy) rats. A second surgical session is needed, in which animals will receive intra-parenchymal injections and be implanted with a catheter to the right jugular vein and left carotid artery. Seven days later, euglycemic-hyperinsulinemic clamp studies will then be performed to measure sensitivity. The reason for this second surgical session is that the selective vagal deafferentation is expected to have small but significant changes on food intake and body weight that will significantly affect insulin sensitivity. Therefore it is necessary to give sufficient time for animals to recover before performing intra-parenchymal injections and implanting catheters. If all surgeries were conducted in one single session, if changes in insulin sensitivity using the clamp were detected, the interpretation of the experiments would be impossible because of the inability to</p>

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	<p>distinguish whether these effects were due to the experimental treatment or because animals simply lost weight after surgery.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Brain cannulation 2. Catheterization of carotid artery and jugular and portal veins, a minimum of 7 days following #1 <p>Species: Rats Interval: 7 days minimum Rationale: To determine the role of the sympathetic nervous system in mediating the central effects of leptin on glucose metabolism, animals will receive an ICV cannula. In addition animals will receive catheters to the jugular vein, carotid artery and portal vein. For some experiments, the surgeries will be separated by at least one week to enable animals to recover from surgery and verify cannula placement. The main reason for the separate surgical sessions is that the patency of the catheters reduces over time and if the surgeries were completed in the same surgical session, a greater percentage of catheters would be blocked and those animals would not be able to be used for the experiment.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Brain cannulation 2. Peripheral parenchymal injections and implantation of osmotic minipump, a minimum of 7 days following #1 <p>Species: Rats Interval: 7 days minimum Rationale: In this study, the aim is to determine the peripheral mechanisms whereby leptin suppresses hepatic glucose production (HGP) in uDM. The lab’s hypothesis is that in uDM, increased sympathetic outflow through sympathetic post-ganglionic neurons located in the celiac ganglion stimulates glucagon release, while increasing sympathetic outflow to the liver, thereby increasing HGP, and this effect is suppressed by leptin action in the brain. This study requires the brain cannulation and the implantation of a mini-pump to be in separate surgical sessions. The reason for this is that the drug being infused by the mini-pump is expected to have small but significant changes on food intake and body weight. Therefore it is necessary to give sufficient time for an animal to recover after the brain cannulation surgery and return to normal food intake and body weight before investigating the actions of the drug administered by mini-pump. If not, it is impossible to distinguish whether changes in feeding or body weight are due to the drug or simply differences in recovery after surgery.</p> <p>or version 174</p> <ol style="list-style-type: none"> 1. Catheterization of carotid artery and jugular veins(to be done by vendor). 2. Brain cannulation at least 7 days post #1 <p>Species: Rats Interval: 7 days minimum Rationale: To determine whether the CNS glucose-lowering effects in uncontrolled diabetes are due to a suppression of hepatic glucose production or an increase in tissue glucose uptake, animals are required to have catheters implanted to the jugular vein and carotid artery and also be implanted with a cannula to the brain. The rationale and justification for reversing the surgical order for this experiment and purchasing rats already bearing catheters to the jugular vein and carotid artery from an approved rodent vendor and subsequently implanting a brain cannula are several fold: 1) It allows us to study a larger cohort of animals with all the necessary controls as one group. This significantly reduces variation when performing these sensitive metabolic studies over two or three groups. We are otherwise limited on the time required to perform the necessary catheter surgeries. 2) It minimizes the potential loss of viable animals due to either complications with the surgical implantation of catheters or the failure to have two successfully patent catheters and 3) Harlan has developed and uses a rounded tip catheter which increases the patency of the catheter and reduces maintenance requirements (please see attached) which previously was a concern. We have validated this from our own experience in catheterized animals from Harlan that did not receive brain surgery.</p> <p>OR (version 192)</p> <ol style="list-style-type: none"> 1. Intracerebroventricular Cannulation
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	<p>2. Vertical Sleeve Gastrectomy and Indwelling Jugular Vein and Carotid Artery Catheter Placement, 7 days after #1 Species: Rats Interval: 7 days Rationale: Indwelling catheters are patent for ~14-21 days after placement. Since animals are allowed to recover for 14 days after VSG surgery, placement at the time of VSG surgery ensures that most catheters will be patent at the time of terminal experiment and euthanasia. Performing icv cannulation at the same time as VSG and catheter placement would prolong anesthesia, be overly stressful for animals, and increase perioperative morbidity and mortality.</p> <p>OR (version 213) 1. Adrenalectomy (done at Harlan prior to shipment) 2. Corticosterone pellet implantation, at least 5 days following #1 3. Brain cannulation, 3-5 days following #2 4. Subcutaneous minipump implantation, at least 7 days following #3 Species: Rats Interval: 3 days minimum Rationale: To determine whether suppression of corticosterone is required for the glucose-lowering effects of leptin in uDM, animals are required to receive a brain cannula, an adrenalectomy and be implanted with a corticosterone pellet and osmotic minipump. After discussing with veterinary services, in order to reduce potential mortality during subsequent surgical procedures, ADX animals will be first implanted with corticosterone pellets (40%) designed to release corticosterone continuously to achieve normal physiological levels. The cannulation and implantation of the minipump is required to be conducted in a separate surgical session. This is to give sufficient time for an animal to recover after the brain cannulation surgery and return to normal food intake and body weight and for the animals to be fully STZ-induced diabetic before treating them with leptin.</p> <p>Multiple Major Surgery: 1. Intraparenchymal injection AND Implantation of indwelling optic fiber for optogenetics 2. Adrenalectomy, a minimum of 7 days following #1 3. Implantation of corticosterone pellet, 7 days following #2 Species: Mice Interval: 7 days minimum Rational: We propose to determine whether stimulation of VMN neurons induces hyperglycemia by increasing corticosterone, glucagon or both. To accomplish this, animals will receive an intraparenchymal injection, followed by implantation of an indwelling optic fiber for optogenetics and allowed to recover for a period (>7 days) to allow the virus to express. Animal's will then receive an adrenalectomy or sham operation, be allowed to recover for 7 days and then be implanted with a physiological replacement dose of corticosterone SQ or control.</p>
2464-06	<p>Multiple Major Surgery: 1. Intracranial injection 2. Gonadectomy + Estradiol Pellet Implant + Body Temperature Monitor, 2 weeks after #1 Species: Mice Interval: 2 weeks Rationale: By separating the surgical sessions, the animals are never under anesthesia longer than 1-2 hours (2 hours if bilateral injection; usually closer to 1 hour). Also, the material that is being intracranially injected is delivering a gene to the animals via a viral construct that requires 3 weeks for full expression in neurons. Thus, it seems preferable to perform one quick invasive procedure, allow a full two weeks for recovery, then perform the other invasive procedure quickly. Though the animals must undergo two procedures where sutures or wound clips are used, it is better to have the animal not recover from all of these invasive procedures at once. Recovery from each procedure individually is very rapid. Since the experimental design allows for this less-stressful course of surgeries, it is preferable to making the animals undergo a potentially 3-hour procedure and longer, more traumatic recovery period.</p> <p>OR</p>

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	<p>Multiple Major Surgery: 1. Intracranial injection 2. SubCue-Mini Datalogger Insertion, 2 weeks after #1 Species: Mice Interval: 2 weeks Rationale: By separating the surgical sessions, the animals are never under anesthesia longer than 1-2 hours (2 hours if bilateral injection; usually closer to 1 hour). Also, the material that is being intracranially injected is delivering a gene to the animals via a viral construct that requires 3 weeks for full expression in neurons. Thus, it seems preferable to perform one quick invasive procedure, allow a full two weeks for recovery, then perform the other invasive procedure quickly. Though the animals must undergo two procedures where sutures or wound clips are used, it is better to have the animal not recover from all of these invasive procedures at once. Recovery from each procedure individually is very rapid. Since the experimental design allows for this less-stressful course of surgeries, it is preferable to making the animals undergo a potentially 3-hour procedure and longer, more traumatic recovery period.</p> <p>OR</p> <p>Multiple Major Surgery: 1. Intracranial injection 2. Gonadectomy + Estradiol Pellet Implant, 2 weeks after #1 Species: Mice Interval: 2 weeks Rationale: By separating the surgical sessions, the animals are never under anesthesia longer than 1-2 hours (2 hours if bilateral injection; usually closer to 1 hour). Also, the material that is being intracranially injected is delivering a gene to the animals via a viral construct that requires 3 weeks for full expression in neurons. Thus, it seems preferable to perform one quick invasive procedure, allow a full two weeks for recovery, then perform the other invasive procedure quickly. Though the animals must undergo two procedures where sutures or wound clips are used, it is better to have the animal not recover from all of these invasive procedures at once. Recovery from each procedure individually is very rapid. Since the experimental design allows for this less-stressful course of surgeries, it is preferable to making the animals undergo a potentially 3-hour procedure and longer, more traumatic recovery period.</p> <p>OR (Version 143)</p> <p>Multiple Major Surgery: 1. Gonadectomy 2. Implantation of SubCue Mini DataLogger, 18 days after #1 Species: Mice Interval: 18 days Rationale: The gonadectomy event will cause the animals' sex steroid level to drop precipitously, but the group needs to provide the animals with enough time to recover and for the levels of T and E to drop completely (21 days). The DataLoggers will be implanted on day 18 to allow a few days of recovery before core body temperatures in sex steroid-free animals are measured on day 25.</p> <p>Multiple Major Surgery: 1. SubCue-Mini Datalogger Insertion 2. Jugular Catheter Implantation, 1 week after #1 3. Repair Catheter (only when fits with repair requirements), at least 3 days after #2 Species: Rats Interval: 3 days to 1 week Rationale: Since both procedures involve inserting a foreign entity into the animal, the animal will encounter less stress and experience optimal recovery when the procedures are separated. Also, the combined time it would take to perform both procedures at once is not as ideal as quickly performing the mini-mitter insertion and then later quickly performing the catheter implantation using an optimally organized surgical station without the added complication of SubCue-Mini Datalogger insertion.</p>
2650-07	Multiple Major Surgery:

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	<p>1. Partial Renal Ablation of Right Kidney 2. Total Left Nephrectomy, 2 weeks following #1 Interval: 2 weeks Species: Mice Rationale: It is necessary to perform a 2-step renal surgery model because the animals will not recover if both surgeries are done at the same time. Two surgeries provides for better survival and better induction of uremia.</p>
2689-01	<p>Multiple Major Surgery: 1. Laparotomy for oocyte harvest 2. Laparotomy for oocyte harvest, minimum of 1 month following #1 3. Laparotomy for oocyte harvest, minimum of 1 month following #2 4. Laparotomy for oocyte harvest, minimum of 1 month following #3 5. Laparotomy for oocyte harvest, minimum of 1 month following #4 6. Terminal laparotomy for oocyte harvest, minimum of 1 month following #5 Species: Xenopus Interval: 1 month minimum Rationale: If a single animal has viable oocytes then multiple surgeries will allow researchers to collect the most data possible per animal.</p>
2856-01	<p>Multiple Major Surgery: 1. ICV injection of AAV vector 2. Middle cerebral artery occlusion 3 Months after #1 Species: Mice Interval: 3 Months Rationale: We need to reduce HDAC expression via shRNA virus injection prior to MCAO procedure.</p>
2950-01	<p>Multiple Major Surgery: 1. Ovariectomy 2. Craniotomy, 5-14 days following #1 Species: Rats Interval: 5-14 days Rationale: Rats must recover for 5-14 days prior to entering study so that hormone levels can normalize.</p>
3055-09	<p>Multiple Major Surgery: Option I 1. MCAO +/- radio telemeter implantation 2. Reperfusion Species: Rat Interval: 2 hours Rationale: The first surgery is MCAO. The second surgery allows for reperfusion and access of the brain tissue by the immune system. It is much safer to allow the animal to recover between these short surgeries than to keep it anesthetized for 2 hours.</p> <p>OR Option II 1. Splenic nerve dissection 2. MCAO +/- radio telemeter implantation 4 days to 2 weeks following #1 3. Reperfusion 2 hours following #2</p> <p>Species: Rat Interval: 2 hours to 2 weeks Rationale: The first surgery is done to prepare for the animal for the experiment - i.e., to prevent sympathetic nerve discharges to the spleen at the time of the stroke. The delay between this splenic nerve dissection and MCAO is to allow the animal to fully recover from surgery and to be certain the spleen is truly denervated. The third surgery is the reperfusion that is necessary to allow blood flow to return to the MCA (and thus the immune system access to the brain tissue). It is much safer to allow the animal to recover between these short surgeries than to keep it anesthetized for 2 hours.</p>
3111-01	<p>Multiple Major Surgery: 1. Ischemia Reperfusion/Permanent ligation of left coronary artery</p>

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	<p>2. Thoracotomy for cell injection/Fibrin scaffold implant (rats only), 5 days to 2 weeks following #1 Species: Mice and Rats Interval: 5 days - 2 weeks Rationale: In many cases it is necessary to perform 2 survival surgeries. These experiments involve one surgery to induce cardiac injury and a second to administer cells/scaffolds directly into the injury during the healing phase.</p> <p>Multiple Major Surgery: 1. Ischemia Reperfusion/Permanent ligation of left coronary artery 2. Thoracotomy for AV injection, 2 to 8 weeks following #1 Species: Mice and Rats Interval: 2 - 8 weeks Rationale: In many cases it is necessary to perform 2 survival surgeries. These experiments involve one surgery to induce cardiac injury and a second to administer AV directly into the injury during the healing phase.</p>
3202-01	<p>Multiple Major Surgery: 1. Orthotopic implantation of the prostate 2. Ablation of the dorsal prostate, a minimum of 3 weeks following #1 Species: Mice Interval: 3 weeks minimum Rationale: These two procedures are required to allow for metastatic sites to grow to the level of detection and provide adequate amounts of tissue for in vitro studies. Without removal of the primary tumor, the health of the animal would be compromised before metastatic sites become macroscopic.</p> <p>OR</p> <p>1. Castration 2. Sub-renal capsule implantation, a minimum of 2 weeks following #1 Species: Mice Interval: 2 weeks minimum Rationale: Castration will be done first followed by sub-renal capsule implantation of tumor tissue. By performing the castration and allowing the testosterone levels to nadir, the tumor won't have the opportunity to revert back to androgen dependence.</p> <p>OR</p> <p>1. Castration 2. Intra-tibial injection OR Subcutaneous implantation, a minimum of 2 weeks following #1 Species: Mice Interval: 2 weeks minimum Rationale: Implanting tumors into already castrated mice will produce androgen independent human prostate cancer xenograft lines.</p> <p>OR</p> <p>1. Intra-tibial injection OR Subcutaneous implantation 2. Castration, a minimum of 1 week following #1 Species: Mice Interval: 1 week minimum Rationale: Performing castration after intra-tibial or subcutaneous injection mimics clinical cases of androgen dependent cancer treatment.</p>
3230-01	<p>Multiple Major Surgery: 1. Craniotomy for fluid percussion injury 2. Headset and ECoG electrode implantation, minimum 3 days following #1 3. Headset/ECoG electrode repair, if needed, minimum 1 week following #2 Species: Rats Interval: 3 days - 1 week minimum Rationale: ECoG electrode implantation following FPI--Later implantation of electrodes in a second surgery is typically necessary in some animals for one of three reasons: 1) Age is a factor that affects</p>

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epileptogenesis in humans. Children are more at risk of post-traumatic epileptogenesis. Also neuronal death is age dependent. We want to study epileptogenesis as it applies to the younger human population. 2) Acute post-injury righting time may be tested as an acute predictor of later epileptogenesis. Therefore, some animals need to emerge from anesthesia after FPI for the righting time to be assessed, in which case electrodes cannot be implanted immediately after FPI. 3) In some experiments we have to balance between the need to start recording EEG as early as possible after injury, with the need to wait so as not to lose the animal's headset. Indeed, the younger the age at which the electrodes get implanted, the more likely the headset will be dislodged as the animal grows. Because part of our studies involve determining whether a correlation exists between acute abnormalities in rat EEG after head injury and the later probability to develop post-traumatic epilepsy, we need to use both FPI + ECoG electrode implantation and FPI + Later ECoG electrode implantation.

ECoG electrode implantation repair--Animals previously implanted with ECoG electrodes may later lose the original headset. This may occur because the animals manage to rip it off. The longer the study post-injury the more likely it is that the rat will manage to rip it off. In our experience, ~20% of animals manage to rip the headset off by 3 months post-injury. If animal lose the headset <3 weeks post-FPI, it will be euthanized because the loss is likely unrecoverable due to the FPI craniotomy having not fully healed. If rats lose the headset >3 weeks after FPI, then the skull has typically healed. The rat will then undergo the ECoG electrode implantation procedure. No more than one re-implantation is ever performed per rat.

OR

1. Craniotomy for fluid percussion injury
2. Headset and ECoG electrode and cooling coil implantation, 12 hrs to 6 weeks following #1
3. Headset/ECoG repair, if needed, minimum 1 week post #2

Species: Rats

Interval: 12hrs - 6 weeks

Rationale: ECoG electrode implantation plus cooling device implantation. Because passive cooling needs to start as early as 12hrs after injury, to simulate treatment of head injury patients in the intensive care unit, the cooling device must also be implanted as early as 12hrs after injury. Since the ECoG electrodes are in the same headset, they also are implanted during the same surgery.

ECoG electrode implantation repair-- Animals previously implanted with ECoG electrodes may later lose the original headset. This may occur because the animals manage to rip it off. The longer the study post-injury the more likely it is that the rat will manage to rip it off. In our experience, ~20% of animals manage to rip the headset off by 3 months post-injury. If animal lose the headset <3 weeks post-FPI, it will be euthanized because the loss is likely unrecoverable due to the FPI craniotomy having not fully healed. If rats lose the headset >3 weeks after FPI, then the skull has typically healed. The rat will then undergo the ECoG electrode implantation procedure. No more than one re-implantation is ever performed per rat.

OR

1. Craniotomy for fluid percussion injury (or sham or naive)
2. Headset and ECoG electrode and cooling coil implant, minimum 12 hrs following #1
3. Cooling piece replacement, minimum of 1 week post #2
4. Cooling piece replacement, minimum of 1 week post #3

Species: Rats

Interval: 12 hrs - 1 week minimum

Rationale: ECoG electrode implantation plus cooling device implantation. Because passive cooling needs to start as early as 12hrs after injury, to simulate treatment of head injury patients in the intensive care unit, the cooling device must also sometimes be implanted as early as 12hrs after injury. Since the ECoG electrodes are in the same headset, they also are implanted during the same surgery.

Cooling device replacement- Since the cooling device cannot be turned on/off, as it cools by heat dissipation, to test its anti-epileptogenic effect we need to remove it from the headset and replace it with an equally sized biocompatible sterile silicone rod that does not dissipate heat. To test the antiepileptic effect, we need to first let the animals become epileptic with a dummy device implanted, then switch the dummy with the cooling device to test the effect, and then switch the cooling device with the dummy rod to acquire a post-cooling baseline. The second baseline is necessary to determine whether seizures return, as expected

<p>for a true antiepileptic effect, and to put the antiepileptic effect of cooling in context with other antiepileptic drugs.</p> <p>OR</p> <ol style="list-style-type: none"> 1. FPI (or sham or naive) + Headset and ECoG electrode implantation 2. Headset and ECoG electrode implantation repair, minimum of 1 week following #1 <p>Species: Rats Interval: 1 week minimum Rationale: ECoG electrode implantation repair-- Animals previously implanted with ECoG electrodes may later lose the original headset. This may occur because the animals manage to rip it off. The longer the study post-injury the more likely it is that the rat will manage to rip it off. In our experience, ~20% of animals manage to rip the headset off by 3 months post-injury. If animal lose the headset <3 weeks post-FPI, it will be euthanized because the loss is likely unrecoverable due to the FPI craniotomy having not fully healed. If rats lose the headset >3 weeks after FPI, then the skull has typically healed. The rat will then undergo the ECoG electrode implantation procedure. No more than one re-implantation is ever performed per rat.</p> <p>OR</p> <ol style="list-style-type: none"> 1. ECoG electrode implantation 2. Headset repair, if needed minimum 1 week after #1 <p>Species: Rats Interval: 1 week minimum Rationale: ECoG electrode implantation repair-- Animals previously implanted with ECoG electrodes may later lose the original headset. This may occur because the animals manage to rip it off. The longer the study post-injury the more likely it is that the rat will manage to rip it off. In our experience, ~20% of animals manage to rip the headset off by 3 months post-injury. If animal lose the headset <3 weeks post-FPI, it will be euthanized because the loss is likely unrecoverable due to the FPI craniotomy having not fully healed. If rats lose the headset >3 weeks after FPI, then the skull has typically healed. The rat will then undergo the ECoG electrode implantation procedure. No more than one re-implantation is ever performed per rat.</p> <p>OR</p> <ol style="list-style-type: none"> 1. FPI 2. ECoG electrode implantation minimum 3 days after #1 3. Headset repair, if needed minimum 1 week from loss of electrodes <p>Interval: Minimum 3 days to 1 week Rationale: ECoG electrode implantation following FPI--Later implantation of electrodes in a second surgery is typically necessary in some animals for one of three reasons: 1) Age is a factor that affects epileptogenesis in humans. Children are more at risk of post-traumatic epileptogenesis. Also neuronal death is age dependent. We want to study epileptogenesis as it applies to the younger human population. 2) Acute post-injury righting time may be tested as an acute predictor of later epileptogenesis. Therefore, some animals need to emerge from anesthesia after FPI for the righting time to be assessed, in which case electrodes cannot be implanted immediately after FPI. 3) In some experiments we have to balance between the need to start recording EEG as early as possible after injury, with the need to wait so as not to lose the animal's headset. Indeed, the younger the age at which the electrodes get implanted, the more likely the headset will be dislodged as the animal grows. Because part of our studies involve determining whether a correlation exists between acute abnormalities in rat EEG after head injury and the later probability to develop post-traumatic epilepsy, we need to use both FPI + ECoG electrode implantation and FPI + Later ECoG electrode implantation. ECoG electrode implantation repair-- Animals previously implanted with ECoG electrodes may later lose the original headset. This may occur because the animals manage to rip it off. The longer the study post-injury the more likely it is that the rat will manage to rip it off. In our experience, ~20% of animals manage to rip the headset off by 3 months post-injury. If animal lose the headset <3 weeks post-FPI, it will be euthanized because the loss is likely unrecoverable due to the FPI craniotomy having not fully healed. If rats lose the headset >3 weeks after FPI, then the skull has typically healed. The rat will then undergo the ECoG electrode implantation procedure. No more than one re-implantation is ever performed per rat.</p>
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	<p>OR</p> <ol style="list-style-type: none"> 1. ECoG electrode plus cooling, or mock cooling device implantation (+Peltier cell in some cases). [Animals that receive the peltier cell will not receive any additional survival surgeries.] 2. Cooling device, or mock cooling device replacement minimum 1 week after #1 3. Cooling device, or mock cooling device replacement [may be repeated once to switch cooling piece, or acquisition of post-cooling baseline.] minimum 1 week after #2. <p>Species: Rats Interval: Minimum 1 week Rationale: ECoG electrode implantation plus cooling device implantation. (+peltier cell in some cases)-- In some cases, to monitor the effects of the cooling on ECoG activity, either a mock cooling device, or cooling device will be implanted on naïve animals. ECoG data will then be acquired. In some cases, in order to see if cooling by more than 2.5 degrees will have a greater impact on the epileptic focus, some animals will receive a peltier cell on the exterior side of the scalp of the animal. This device will allow us to cool the brain area of interest by up to 6oC which has been shown to be safe (Rothman et al 2005). If the peltier is gets sloughed or loosened it is re-secured using vet-bond adhesive. The peltier cell is removed at the end of the study. The removal of the peltier cell occurs at time of euthanasia of the animal. Cooling device replacement- In animals that do not receive the peltier cell, the cooling device cannot be turned on/off, as it cools by heat dissipation. To test its anti-epileptogenic effect we need to remove it from the headset and replace it with an equally sized biocompatible sterile silicone rod that does not dissipate heat. To test the antiepileptic effect, we need to first let the animals become epileptic with a dummy device implanted, then switch the dummy with the cooling device to test the effect, and then switch the cooling device with the dummy rod to acquire a post-cooling baseline. The second baseline is necessary to determine whether seizures return, as expected for a true antiepileptic effect, and to put the antiepileptic effect of cooling in context with the one of the other antiepileptic drugs.</p> <p>OR</p> <ol style="list-style-type: none"> 1. FPI 2. ECoG electrode and mock cooling, or cooling device implantation (+Peltier cell in some cases) Animals that receive the peltier cell will not receive any additional survival surgeries. Minimum 12 hr after #1. 3. Cooling Device replacement. Minimum 1 week after #2. 4. Cooling Device replacement (may be repeated once to switch cooling device, or acquisition of post-cooling baseline.) Minimum 1 week after #3. <p>Species: Rats Interval: Minimum 12 hr to 1 week Rationale: ECoG electrode implantation plus cooling device implantation.(+peltier cell in some cases)-- Later implantation of electrodes in a second surgery is typically necessary in some animals for one of three reasons: 1) Age is a factor that affects epileptogenesis in humans. Children are more at risk of post-traumatic epileptogenesis. Also neuronal death is age dependent. We want to study epileptogenesis as it applies to the younger human population. 2) Acute post-injury righting time may be tested as an acute predictor of later epileptogenesis. Therefore, some animals need to emerge from anesthesia after FPI for the righting time to be assessed, in which case electrodes cannot be implanted immediately after FPI. 3) In some experiments we have to balance between the need to start recording EEG as early as possible after injury, with the need to wait not to lose the animal's headset. Indeed, the younger the age at which the electrodes get implanted, the more likely the headset will be dislodged as the animal grows. Because part of our studies involve determining whether a correlation exists between acute abnormalities in rat EEG after head injury and the later probability to develop post-traumatic epilepsy, we need to use both FPI + ECoG electrode implantation and FPI + Later ECoG electrode implantation. In some cases, since passive cooling needs to start as early as 12hrs after injury, to simulate treatment of head injury patients in the intensive care unit, the cooling piece, which works passively by dissipating heat without power, must also be implanted as early as 12hrs after injury. In some cases, at the time of electrode and cooling device implantation, in order to see if cooling by more than 2.5 degrees will have a greater impact on the epileptic focus, some animals will receive a peltier cell</p>
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<p>on the exterior side of the scalp of the animal. This device will allow us to cool the brain area of interest by up to 6°C which has been shown to be safe (Rothman et al 2005). If the peltier is gets sloughed or loosened it is re-secured using vet-bond adhesive. The peltier cell is removed at the end of the study. The removal of the peltier cell occurs at time of euthanasia of the animal.</p> <p>Cooling device replacement- In animals that do not receive the peltier cell the cooling device cannot be turned on/off, as it cools by heat dissipation, to test its anti-epileptogenic effect we need to remove it from the headset and replace it with an equally sized biocompatible sterile silicone rod that does not dissipate heat. To test the antiepileptic effect, we need to first let the animals become epileptic with a dummy device implanted, then switch the dummy with the cooling device to test the effect, and then switch the cooling device with the dummy rod to acquire a post-cooling baseline. The second baseline is necessary to determine whether seizures return, as expected for a true antiepileptic effect, and to put the antiepileptic effect of cooling in context with other antiepileptic drugs.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Cooling Device replacement. 2. Cooling Device replacement. (May be repeated once to switch cooling device.) Minimum 1 week after #1. <p>Species: Rats Interval: Minimum 1 week</p> <p>Rationale: Cooling device replacement- Since the cooling device cannot be turned on/off, as it cools by heat dissipation, to test its anti-epileptogenic effect we need to remove it from the headset and replace it with an equally sized biocompatible sterile silicone rod, or acrylic mock cooling rod, that does not dissipate heat. To test the antiepileptic effect, we need to first let the animals become epileptic with a mock cooling device implanted, then switch the mock cooling device with the cooling device to test the effect, and then switch out the cooling device with a mock cooling device to acquire a post-cooling baseline. The second baseline is necessary to determine whether seizures return, as expected for a true antiepileptic effect, and to put the antiepileptic effect of cooling in context with other antiepileptic drugs.</p> <p>OR</p> <ol style="list-style-type: none"> 1. FPI plus Cooling Device implantation. 2. Cooling Device replacement. (May be repeated once to switch cooling device.) Minimum 3 days after #1. <p>Species: Rats Interval: Minimum 3 days</p> <p>Rationale: Cooling device replacement- Since the cooling device cannot be turned on/off, as it cools by heat dissipation, to test its anti-epileptogenic effect we need to remove it from the headset and replace it with an equally sized biocompatible sterile silicone rod that does not dissipate heat. To test the antiepileptic effect, we need to first injure the animals and let them become epileptic with a mock cooling device implanted. Then we need to switch the mock cooling device with the cooling device to test the effect of cooling. Then we need to switch the cooling device with mock cooling device to acquire a post-cooling baseline. The second baseline is necessary to determine whether seizures return, as expected for a true antiepileptic effect, and to put the antiepileptic effect of cooling in context with other antiepileptic drugs.</p> <p>OR</p> <ol style="list-style-type: none"> 1. FPI 2. ECoG Electrode Implanation minimum 3 days after #1 3. Osmotic mini-pump implantation minimum 3 days from #2 In some cases surgeries 2 and 3 may be done in the same surgical session. 4. Osmotic mini-pump removal/replacement minimum 3 days after #3. <p>Species: Rats Interval: Minimum 3 days</p> <p>Rationale: ECoG electrode implantation following FPI--Later implantation of electrodes in a second surgery is typically necessary in some animals for one of three reasons: 1) Age is a factor that affects epileptogenesis in humans. Children are more at risk of post-traumatic epileptogenesis. Also neuronal death is age dependent. We want to study epileptogenesis as it applies to the younger human population. 2) Acute post-injury righting time may be tested as an acute predictor of later epileptogenesis. Therefore,</p>
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	<p>some animals need to emerge from anesthesia after FPI for the righting time to be assessed, in which case electrodes cannot be implanted immediately after FPI. 3) In some experiments we have to balance between the need to start recording EEG as early as possible after injury, with the need to wait so as not to lose the animal's headset. Indeed, the younger the age at which the electrodes get implanted, the more likely the headset will be dislodged as the animal grows. Because part of our studies involve determining whether a correlation exists between acute abnormalities in rat EEG after head injury and the later probability to develop post-traumatic epilepsy, we need to use both FPI + ECoG electrode implantation and FPI + Later ECoG electrode implantation.</p> <p>To study the effect of antiepileptic drugs on FPI-induced seizures, drugs have to be delivered. In some cases to maintain stable blood levels of drug, s.c.u osmotic mini-pumps must be used. Pumps are installed from a minimum 3 days after injury. This same procedure of pump implantation may be repeated once after a minimum 1 week from initial pump implantation to either removed or replace the pump. In the case in which immediate stoppage of drug delivery is needed, the pump may be removed. In the case in which a pump needs to be replaced in order to continue drug delivery the pump may be replaced. All surgical sessions will be separated by at least 3 days.</p>
3441-05	<p>Multiple Major Surgery: 1. Laparotomy, Hepatic subcapsular tumor injection 2. Laparotomy, Hepatic artery infusion pump 1-3 weeks after #1 Species: Rats Interval: 1-3 weeks Rationale: 1 to 3 weeks between surgeries is necessary to ensure that the laparotomy was successful before proceeding with the pump surgery</p> <p>Multiple Major Surgery: 1. Intracranial tumor xenograft induction 2. Cranial implantation of Cannula Species: Mice Interval: 7-21 days Rationale: Intracranial tumor xenograft induction promotes the growth of tumor cells within the cranium. This process yields a more realistic brain tumor model to address the difficulties associated with current clinical methods of imaging and treatment of brain tumors. Cranial cannula implantation is necessary to facilitate serial convection enhanced delivery events. 7-21 days between tumor implantation and cranial cannula implantation is necessary to ensure that the intracranial tumor xenograft was successfully established before proceeding with convection enhanced delivery via cannula infusion.</p>
4053-01	<p>Multiple Major Surgery: 1. Minipump IP implant 2. Minipump IP explant Species: Mice Interval: 10 days Rationale: Exhausted pumps continue to absorb fluid and begin to leak concentrated salts after the reservoir is empty, so removal is recommended by manufacturer.</p>
4073-01	<p>Multiple Major Surgery: 1. Intracerebral microinjector cannulation, injectrode or voltammetry electrode implantation (with or without intracranial microinfusion of AAV vectors) 2. Intravenous (jugular) catheter implantation, 1-4 weeks after #1 Species: Rats Interval: 1-4 weeks Rationale: Indwelling jugular catheter surgeries will be performed separately from intracranial implantation surgeries a) to minimize the total time of anesthesia per surgery, and b) because the longevity of catheter patency is limited and thus needs to be performed in closer proximity to the behavioral studies. To allow for adequate recovery from the intracranial implantation surgery, catheter surgeries will be performed at least 1 week and up to 4 weeks after intracranial implantation surgery.</p>
4079-01	<p>Multiple Major Surgery: 1. Steroid implant procedure 2. Auditory physiology recordings, 23-35 days following #1</p>

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	<p>Species: Fish Interval: 23-35 days Rationale: Multiple surgical sessions are required to test for the effects of steroids on auditory physiology.</p>
4105-01	<p>Multiple Major Surgery: 1. Intracranial injection of tumor 2. Canular implantation for drug injection Species: Mouse Interval: 5-7 days Rationale: Intial surgery will be establish a brain tumor in the mouse. the second surgery is to implant a cannula for drug/mono-clonal antibody injections into the tumor cavity.</p>
4133-01	<p>Multiple Major Surgery: 1. Headpost Implantation 2. Chamber 1 implantation 3. Craniotomy for chamber 1 4. Craniotomy 1 expansion 5. Chamber 2 implantation 6. Craniotomy for chamber 2 7. Craniotomy expansion 8. Chamber 3 implantation 9. Craniotomy for chamber 3 10. Craniotomy 3 expansion 11. Chamber 4 implantation 12. Craniotomy for chamber 4 13. Craniotomy 4 expansion 14. Array Implantation 15. CTB injections 16. Potential repair surgery #1, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment 17. Potential repair surgery #2, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment Species: M. mulatta Interval: 1-3 months minimum Rationale: Our experiments require a minimum of 3 surgeries to obtain any experimental data using the standard chamber method – implantation of a head post, implantation of a recording chamber and a craniotomy. These three surgeries are incompatible to being performed as one because, the head post surgery requires a recovery period of 6 weeks, while a craniotomy requires that we start experiments the very next day. If using the array method, our experiments require a minimum of 2 surgeries to obtain any experimental data – implantation of a headpost and implantation of the array which are incapable of being done as a single surgery for the same reasons as with the chamber surgeries. However, these surgeries are not sufficient for successfully achieving our research goals due to three reasons: First, when the goal of research is to understand higher order brain function, it is important to compare the roles of multiple different brain areas in the same animal, since brain areas seldom operate in isolation to effect higher order cognitive tasks. This requires the placement of multiple recording chambers. Secondly, when the areas of interest are on the surface of the cortex, it is important for the dural surface to be fresh for successful recordings. This necessitates that we perform the i) craniotomy immediately prior to experiments to increase the success ii) and to expose small regions of the cortex periodically to facilitate an extensive period of successful data collection. Thirdly, we make a huge investment in time and effort to prepare these animals for recording – animals typically spend a year to 1.5 years getting trained on a behavioral task – replacing this animal with another naïve animal (because experimental hardware came undone, etc) would be a loss of valuable effort and time. Repair Surgeries: We request 2 repair surgeries in the event there is need for a repair or correction. This applies to animals</p>

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	<p>new to the protocol only. It does not apply to carry-over animals since their surgery/ies are already approved by the IACUC. Additional repair surgeries require IACUC approval, which will be requested through a Significant Change. This is in accordance with the UW Guidelines of Non-Human Primate Neuroscience Studies.</p>
4133-02	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Head post implantation 2. Craniotomy+Injection (Hemisphere 1), at least 3 months following #1 3. Visual Cortex chamber implant, craniotomy and durotomy, at least 3 weeks following #2 4. Craniotomy+Injection (Hemisphere 2), at least 2 months following #3 5. Visual Cortex chamber implant, craniotomy and durotomy (2nd hemisphere) , at least 3 weeks following #4 6. Potential repair surgery #1, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment 7. Potential repair surgery #2, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment <p>Species: M. mulatta Interval: At least 3 weeks up to at least 3 months</p> <p>Rationale: Our experiments require a minimum of 3 surgeries to obtain any experimental data – implantation of a head post, injection of GCaMP or GFP, implantation of a recording chamber with a craniotomy and durotomy. These three surgeries are incompatible to being performed as one because, the head post surgery requires a recovery period of 6 weeks, while the injection need at least 3 weeks for expression, and the chamber, craniotomy and durotomy surgery requires that we start experiments within 2 weeks of implantation.</p>
4146-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Eye coil and lug implantation 2. Chamber implantation, at least 1 month after #1 3. Vestibular prosthesis implantation, at least 1 month after #2 4. Myringotomies (2), at least 1 week after #3 5. Myringotomies (2), at least 1 week after #4 6. Myringotomies (2), at least 1 week after #5 7. Myringotomies (2), at least 1 week after #6 8. Myringotomies (2), at least 1 week after #7 9. Myringotomies (2), at least 1 week after #8 10. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment <p>Species: M. Mulatta Interval: 1 month</p> <p>Rationale: There are three planned survival surgeries. Recovery following the eye coil surgery is a minimum of one week depending on swelling around the orbit. The animals are then trained in the task, which takes from a week to a few months, depending on the difficulty of the task. The animals are then scheduled for a second surgery, typically a chamber surgery. After the animals have recovered, we then map the chamber(s) and schedule a third surgery, typically a vestibular implant surgery. Occasionally, we will reverse the order, and perform the vestibular implant surgery second, depending on the schedule for recording single neurons. We use three surgeries because recovery is much quicker when the animals are not anesthetized for periods of greater than a few hours and because the stability of the implants is improved when the anchoring screws for the stabilization lugs are allowed to integrate more completely prior to the craniotomy for the chamber(s). The minimum time between surgeries is 1 month. It is the interval that it takes to train the animal to track the spot under all test conditions following implant of the coil, and become comfortable with working in the testing environment of the laboratory. The interval depends on how quickly the monkey acquires the behavioral task and how complex the task is. In our experience, it has taken monkeys at least a month to learn to perform simple tracking task for 2-3 hours. We do not plan to have to repair or adjust our implants, but if we do, such a repairs would be more likely to occur after several months of recording. WE WOULD SUBMIT A MODIFICATION REQUEST TO THE</p>

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	<p>IACUC FOR ANY REPAIR SURGERY FOLLOWING THE FIRST REPAIR. FURTHERMORE, ALL SURGERIES, INCLUDING REPAIR SURGERIES, REQUIRE A PRESURGICAL EXAMINATION, VETERINARY REVIEW OF ALL RELEVANT ANIMAL HEALTH RECORDS, AND VETERINARY AUTHORIZATION PRIOR TO SURGERY. Chamber adjustments do not require additional craniotomies. In our experience, chamber adjustments occur in less than 1/10 animals, and lug or post replacement or repair occurs with a frequency of 1/ 2 animals.</p> <p>Surgeries 4-9 (serial titrated injections of pharmaceutical grade gentamicin) A single surgical procedure with higher gentamicin concentrations would be too stressful for the animals and may compromise hearing function. There are multiple planned procedures to titrate the effect of the injections. We will continue injecting the ears until we have at least a 70% reduction in function bilaterally. By performing serial injections, we avoid a rapid acute loss of vestibular function, resulting in whirling vertigo, or a loss of hearing function. These procedures are well tolerated in humans and monkeys, and are a direct analog of outpatient procedures performed clinically during regular office visits in humans.</p>
4154-01	<p>Multiple Major Surgery: 1. Transient middle artery occlusion 2. ICV injection, 24-48 hours following #1 3. Middle artery reperfusion, 24 hours following #2 Species: Mice Interval: between 1 and 2 24-48 hours Interval: between 2 and 3 24 hours Rationale: Ischemic preconditioning requires genomic reprogramming and gene expression. Classically, these changes require 48-72 hours to take effect and last several weeks. These are the earliest time point possible (48-72 hours) for scientific/experimental design reasons and in order to minimize post-surgical time for the animal.</p>
4165-01	<p>Multiple Major Surgery: 1. Maternal-fetal vascular catheterization, myometrial and membrane biopsies (see V54 for slight change) 2. C-section, myometrial and membrane biopsies, minimum of 10 days following #1 Species: <i>Macaca nemestrina</i> Interval: minimum of 10 days Rationale: The second major survival surgery is performed in order to obtain fetal and placental tissues under controlled conditions. If the dam is allowed to deliver vaginally, the fetus may die during the birth, rendering experimental results from the fetal tissues difficult to interpret. If the delivery occurs at night, the dam often ingests the placental tissues, preventing scientific use of these tissues.</p> <p>Multiple Major Surgery (version 53): 1. Maternal-fetal vascular catheterization #1 2. C-section #1, at least 10 days following #1 3. Maternal-fetal vascular catheterization, #2, at least 180 days following #2 (Surgery 2 for all animals mentioned in this significant change was at least 180 days ago.) 4. C-section #2, at least 10 days following #3 Species: <i>Macaca nemestrina</i> Interval: minimum of 10 days Rationale: The second major survival surgery is performed in order to obtain fetal and placental tissues under controlled conditions. If the dam is allowed to deliver vaginally, the fetus may die during the birth, rendering experimental results from the fetal tissues difficult to interpret. If the delivery occurs at night, the dam often ingests the placental tissues, preventing scientific use of these tissues. Scientific justification for 3rd and 4th surgeries: Rare resources and a diminishing supply of <i>Macaca nemestrina</i>. The goal of my protocol is to study the biology and test new therapies for preterm labor. This is one of only two centers in the world that can perform experiments in a complex chronically catheterized nonhuman primate model of pregnancy. The primate center cannot support our project needs for pregnant animals at this time due to a lack of breeders and need to increase infants supplied to the colony. Our prior experiments have been done in <i>Macaca nemestrina</i> and we would like to continue doing experiments in this species to allow for comparisons across our experiments with prior data.</p>
4167-01	<p>Multiple Major Surgery: 1. Implant of head post and eye coil</p>

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	<p>2. Craniotomy and implantation of recording device #1, minimum of 6 months following #1. 3. Craniotomy and implantation of recording device #2, minimum of 1 year following #2. 4. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment.</p> <p>Species: <i>Macaca mulatta</i> Interval: At least 6 months</p> <p>Rationale: The eye coil and head post are implanted in a single surgical session and the first recording device is implanted during a subsequent surgical session. The justification for not including the first recording device in the first surgical session arises from consideration of the timing of the behavioral training and the desire to guard animal health. Specifically, the animal is prepared for behavioral training as soon as the eye coil and head post have been implanted. Training can take up to a year. During this time, invasive manipulations are not made, obviating the necessity for recording chambers, craniotomies, or multielectrode arrays. Indeed, these hold the potential to compromise the health of the animal: A recording cylinder or multielectrode array connector increases the area of the cranial implant, thereby increasing the likelihood of osteopathy, and a craniotomy raises the risk of intracranial infection. For these reasons we do not generally implant the recording device nor make the craniotomy until after behavioral training is completed. If the behavioral task is sufficiently simple (i.e. ocular fixation) such that training is expected to take less than two months we may implant the recording device during the same surgical session as the eye coil and head post. The second recording device is intended as a replacement for the first recording device. It is not implanted until the first one fails. Implantation of a second recording device allows us to increase the amount of data we can obtain from each animal, thereby reducing the total number of animals used in the completion of the project aims.</p> <p>OR Version #62 (2 approved <i>M. mulatta</i> ONLY)</p> <p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Implant of head post and eye coil 2. Craniotomy and implantation of two recording devices (area V1, bilateral), minimum of 6 months following #1. 3. Craniotomy and implantation of recording device (area SC) and removal of one of the V1 chambers, minimum of 6 months following #2. 4. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Species: <i>Macaca mulatta</i> Interval: At least 6 months</p> <p>Rationale: The eye coil and head post are implanted in a single surgical session and the first recording device is implanted during a subsequent surgical session. The justification for not including the first recording device in the first surgical session arises from consideration of the timing of the behavioral training and the desire to guard animal health. Specifically, the animal is prepared for behavioral training as soon as the eye coil and head post have been implanted. Training can take up to a year. During this time, invasive manipulations are not made, obviating the necessity for recording chambers, craniotomies, or multielectrode arrays. Indeed, these hold the potential to compromise the health of the animal: A recording cylinder or multielectrode array connector increases the area of the cranial implant, thereby increasing the likelihood of osteopathy, and a craniotomy raises the risk of intracranial infection. For these reasons we do not generally implant the recording device nor make the craniotomy until after behavioral training is completed. If the behavioral task is sufficiently simple (i.e. ocular fixation) such that training is expected to take less than two months we may implant the recording device during the same surgical session as the eye coil and head post. The second recording device is intended as a replacement for the first recording device. It is not implanted until the first one fails. Implantation of a second recording device allows us to increase the amount of data we can obtain from each animal, thereby reducing the total number of animals used in the completion of the project aims.</p> <p>OR version 71: Multiple Major Surgery:</p>
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	<p>1. Implant of head post and eye coil 2. Recording device implantation #1, (possibly 2 chambers), minimum of 6 months following #1. 3. Recording device implantation #2, (one chamber-maximum of 2 simultaneously), minimum of 6 months following #2. 4. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. Species: <i>Macaca mulatta</i> Interval: At least 6 months Rationale: The eye coil and head post are implanted in a single surgical session and the first recording device is implanted during a subsequent surgical session. The justification for not including the first recording device in the first surgical session arises from consideration of the timing of the behavioral training and the desire to guard animal health. Specifically, the animal is prepared for behavioral training as soon as the eye coil and head post have been implanted. Training can take up to a year. During this time, invasive manipulations are not made, obviating the necessity for recording chambers, craniotomies, or multielectrode arrays. Indeed, these hold the potential to compromise the health of the animal: A recording cylinder or multielectrode array connector increases the area of the cranial implant, thereby increasing the likelihood of osteopathy, and a craniotomy raises the risk of intracranial infection. For these reasons we do not generally implant the recording device nor make the craniotomy until after behavioral training is completed. If the behavioral task is sufficiently simple (i.e. ocular fixation) such that training is expected to take less than two months we may implant the recording device during the same surgical session as the eye coil and head post. The maximum number of recording devices that a single animal will have simultaneously is two. These two devices will either be implanted during a single surgical session or in two separate sessions. Other animals will have only a single recording device.</p>
4187-02	<p>Multiple Major Surgery: 1. Skull Implant 2. Muscle Implant* (in some cases combined with Skull Implant), at least 2 weeks following #1 3. Spinal Injury* (in some cases combined with Spinal Implant), at least 10 days following #2 4. Spinal Implant*, at least 2 weeks following #3 5. Brain tracer injections*, at least 10 days following #5, or, per version 45, with brain/muscle implant procedures. 6. Repair Surgery (if needed - veterinary services will be consulted prior to performing any repair surgery (per VVC version 49))* Species: Rats Interval: 10 days - 2 weeks minimum Rationale: Multiple surgeries are required to test the ability of different applications of intraspinal electrical stimulation to restore motor function following spinal cord injury. The initial skull surgery must be separated from the remaining surgeries in order to determine the effects of brain recording and/or stimulation prior to injury of the spinal cord. The experimental protocol will compare the effects of starting stimulation soon after injury and several weeks after injury (acute vs. chronic injury conditions). To prevent possible malfunction of the spinal implant between implant and use in the chronic treatment groups the spinal injury and spinal implant procedures will be performed in different surgeries in these animals. The brain tracer must be injected after recovery has taken place due to our experimental intervention. During these long-term studies, it may be occasionally necessary to add sutures or acrylic to protect wires entering the skull implant after the skin has relaxed or eroded. Head caps also become loose over time, and it may be necessary to add several skull screws and new acrylic in a surgical procedure. We therefore request one potential repair surgery for each animal to cover these possibilities. Note that we will not re-approach the spinal cord in these procedures. Repairs will only involve the skin, skull, and superficial muscle layers. *These surgeries will not be performed in some animals.</p>
4187-05	<p>Multiple Major Surgery: 1. Spinal and brain implant 2. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. Species: Macaques</p>

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	<p>Interval: Interval depends on need. Rationale: A repair surgery is performed if the scientific goals of the study have not yet been completed in an individual animal. This is in accordance with the UW Guidelines of Non-Human Primate Neuroscience Studies.</p>
4189-03	<p>Multiple Major Surgery: 1. Injection of rAAV-GAD65 constructs or monoclonal GAD65Ab into the lateral ventricles of the brain. 2. Implantation of EEG electrodes Species: Mice Interval: 3 weeks Rationale: The mice will be first injected with the reagent in the lateral ventricle. After the protein is expressed in the brain, the effect on seizure susceptibility will be evaluated by EEG-recording. Therefore we need to implant the EEG-electrodes after administration of the reagent and protein expression. The second surgery will be only performed in those animals that were injected in the lateral ventricles.</p>
4214-01	<p>Multiple Major Surgery: 1. Coronary artery ligation without occlusion 2. Closed chest myocardial ischemia-reperfusion injury, 1 week following #1 Species: Mice Interval: 1 week Rationale: Closed-chest mouse model of IR allows ligation of the LAD coronary artery posterior to the acute surgical trauma providing a unique opportunity to study the structure, function and energetic metabolism of the mouse heart by MRI/MRS after IR injury in vivo.</p>
4221-01	<p>Multiple Major Surgery: Surgery Table for Groups II, IIIa, IIIb. 1. Eye muscle surgery (possible growth factor placement) 2. Second eye muscle surgery in first month of life or second Growth factor pellet implantation on extraocular muscle (if only 1 eye was done in surgery #1), 1 week to 1 month following #1 3. Eye muscle surgery >1.5 years of age – not all animals receive surgical strabismus and would skip to surgery #4, >1.5 years following #1 or #2 4. Restraining hardware, recording chamber, scleral search eye coil implantation in one eye, or no eye coil placement with both eye coils placed in next surgery, > 2 weeks following #3 5. Scleral search eye coil implantation in second eye, or eye coil in both eyes, > 2 weeks following #4 6. Scleral search eye coil implanted in second eye if only one implanted in earlier surgeries, > 2 weeks following #5 7. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment Species: Macaques Interval: 2 weeks – 1.5 years Rationale: We have developed effective animal models for strabismus using surgical methods like those used in human infants. To achieve our goals, surgical adjustment of eye muscles must be made during infancy (critical period for visual-oculomotor development). For some eye muscle surgery cases, we may treat muscles with growth factors. Because the growth factor is delivered to specific muscles it requires a surgical procedure. When surgical animals reach at least 1.5 years of age, they will be subjects in studies of neural control of eye movements as described under our juvenile protocols. This is where the 3rd surgical procedure is necessary. We need to implant single unit recording chambers over oculomotor regions of the brain to permit neurophysiological studies. We usually perform the juvenile surgery consisting of a coil implant, chambers and head stabilization post all in one surgery. Even an experienced team needs 4-6 hours to accomplish all of this. Therefore, we perform a second scleral search coil to reduce the trauma to the monkey. In rare cases, only the chambers and restraint hardware get placed together. The scleral search coils are implanted separately in one or two surgery sessions. Two can be required depending on the different morphological variation in individual animals. Some animals are significantly more difficult to implant than others. Our scleral search coil implants are minimally disruptive to the animal. Placing the second coil two weeks or more after the first allows the monkey to recover optimally. In some case we implant scleral search coil in one or both eyes first and then after more than 2 weeks, we implant chambers and head post.</p>

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	<p>Sometimes, hardware implanted on the head may need to be reinforced or replaced due to unforeseen circumstances. We request one-repair surgery to be used for these situations. As per the “UW Guidelines of Non-Human Primate Neuroscience Studies” policy we acknowledge that any additional repair/correction surgeries require review and approval by the IACUC.</p> <p>OR</p> <p>Multiple Multiple Surgery Table: Surgery Table for Groups Ic, Id.</p> <ol style="list-style-type: none"> 1. Eye muscle surgery (>1.5 years of age – not all animals receive Surgical Strabismus and would start at Surgery #2.) 2. Restraining hardware, recording chamber, scleral search eye coil implantation in one eye if time allots, > 2 weeks following #1 3. Scleral search coil implantation of one or two coils -OR- Implantation of the second coil, > 2 weeks following #2 4. Scleral search eye coil implanted in second eye if only one implanted in earlier surgeries, > 2 weeks following #3 5. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Note: The order of surgeries 4, 5 & 6 could vary – e.g. we might choose to implant one or two search coils first. Then recording chambers and restraint hardware. Time between these surgeries is always >2 weeks.</p> <p>Species: Macaques Interval: 1 week - 2 years</p> <p>Rationale: We have developed effective animal models for strabismus using surgical methods like those used in human infants. To achieve our goals, surgical adjustment of eye muscles must be made during infancy (critical period for visual-oculomotor development).</p> <p>For some eye muscle surgery cases, we may treat muscles with growth factors. Because the growth factor is delivered to specific muscles it requires a surgical procedure.</p> <p>When surgical animals reach at least 1.5 years of age, they will be subjects in studies of neural control of eye movements as described under our juvenile protocols. This is where the 3rd surgical procedure is necessary. We need to implant single unit recording chambers over oculomotor regions of the brain to permit neurophysiological studies.</p> <p>We usually perform the juvenile surgery consisting of a coil implant, chambers and head stabilization post all in one surgery. Even an experienced team needs 4-6 hours to accomplish all of this. Therefore, we perform a second scleral search coil to reduce the trauma to the monkey. In rare cases, only the chambers and restraint hardware get placed together. The scleral search coils are implanted separately in one or two surgery sessions. Two can be required depending on the different morphological variation in individual animals. Some animals are significantly more difficult to implant than others. Our scleral search coil implants are minimally disruptive to the animal. Placing the second coil two weeks or more after the first allows the monkey to recover optimally. In some case we implant scleral search coil in one or both eyes first and then after more than 2 weeks, we implant chambers and head post.</p> <p>Sometimes, hardware implanted on the head may need to be reinforced or replaced due to unforeseen circumstances. We request one-repair surgery to be used for these situations. As per the “UW Guidelines of Non-Human Primate Neuroscience Studies” policy we acknowledge that any additional repair/correction surgeries require review and approval by the IACUC.</p> <p>OR</p> <p>Surgery Table for Groups Ib (NOT MAJOR)</p> <ol style="list-style-type: none"> 1. Intra-vitreous photo switch injection in one or both eyes 2. Intra-vitreous photo switch injection in second eye, > 2 weeks after #1
4246-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Craniotomy (EEG, thermistor, ICV guide cannula) 2. Cecal Ligation Puncture, 1 week following #1 <p>Species: Mice Interval: 1 week</p>

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	<p>Rationale: Some experiments require sleep recordings to be obtained from animals subjected to CLP and they first require baseline readings prior to CLP.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Telemeter Implant - Laparotomy 2. Cecal Ligation Puncture, 2-4 weeks following #1 <p>Species: Mice Interval: 2-4 weeks</p> <p>Rationale: Some experiments require sleep recordings to be obtained from animals subjected to CLP and they first require baseline readings prior to CLP.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Telemeter Implant - Laparotomy 2. Craniotomy for ICV guide cannula implant, 2-4 weeks following #1 <p>Species: Mice Interval: 2-4 weeks</p> <p>Rationale: The recovery from the telemeter implant is longer than the average patency of an ICV guide cannula.</p>
4253-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Induction of myocardial infarction 2. Engraftment of hiPSC-CMs 4 days later <p>Species: Rats Interval: 4 days</p> <p>Rationale: The separation of the 2 surgeries is important to replicate the typical clinical intervention in the hospital setting. Specifically, the infarction is induced, similar to a patient presenting with a heart attack. The therapeutic cells are introduced into the heart 4 days later, during the healing phase following the initial injury, rather than immediately following the experimentally induced coronary occlusion.</p>
4254-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Titanium restraining hardware and recording chamber (option 1) or along with scleral search coil implant (option 2) 2. Scleral search coil implantation for two eyes (option 1) or second eye (option 2), at least 2 weeks after #1 3. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment <p>Species: <i>M. mulatta</i> Interval: 2 weeks minimum</p> <p>Rationale: We need to implant recording chambers and scleral search coils (both eyes). We implant head posts and recording chambers in one surgery taking 4-5 hours. If time allows, we also place one eye coil, but a surgery to implant both eye coils would be too long for the monkey. In Surgery #2 we implant either one or two eye coils as needed. A broken eye coil might need to be replaced. Stabilization hardware and recording chambers can become loose or break requiring replacement.</p>
4259-03	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Laparotomy for oocyte collection in adult females 2. Laparotomy for oocyte collection in adult females 3. Laparotomy for oocyte collection in adult females <p>Species: Macaca Interval: Minimum of 3 months 1 and 2 Interval: Minimum of 3 months 2 and 3</p> <p>Rationale: In an effort to maximize the yield of oocytes available from an individual female and reduce the total number of animals required over the course of a study up to a maximum of three recovery surgeries are requested. In a single donor cycle the yield of oocytes is based upon the cohort of antral follicles that have been recruited during the previous luteal phase and cannot be exogenously manipulated. In order to utilize this renewable source of oocyte material it is necessary to perform multiple stimulation cycles and subsequent surgeries. Evidence from the literature has shown that females are able to respond to</p>

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	<p>multiple stimulation (3-10 cycles) with exogenous hormones (rhFSH and hCG) without adverse effects on either oocyte number, oocyte quality or subsequent breeding performance (both natural and assisted; Yang et al 2008; VandeVoort & Tarantal, 2001). Under advisement from the Oregon National Primate Research Center where they routinely perform 3-5 ovarian stimulation cycles on the Rhesus macaque with no/minimal adverse outcomes (ie. low response, formation of adhesions). Primarily the limitation in number is due to the use of human hormones and the potential development of antibodies against these agents. The appearance of antibodies does not impact the animal but can reduce the stimulation response. With any surgery the potential for adhesions to develop exists. Under our current practices we take measures to minimize the occurrence of adhesions associated with laparotomy through the use of powder-free and pre-rinsed surgical gloves and lavage of the abdomen to remove blood clots at the conclusion of the procedure. Furthermore, we will be advised by our veterinary surgeons to make the decision of limiting further surgery should appearance adhesions in an individual female be observed. IN support of our request for multiple survival surgeries It should be noted that i)the WaNPRC has exceptional animal identification and record keeping to ensure that each female does not receive more than the maximum number of recoverable surgeries. ii) Surgeries are performed by trained persons using appropriate anesthesia, aseptic technique and pre-, peri- and postoperative analgesia. iii) Single housing after surgery is provided as part of the post-surgical care of laparotomized animals. iv) Females are monitored daily during the postoperative period (at least one week) for appetite as well as any complications such infection. v) Adequate recovery time will be in place between laparotomies (no less than 3 months). vi) The number of laparotomies on an individual female to obtain oocytes will be conditional upon the relative health of an individual animal and quality of the oocytes obtained from the previous collection.</p>
4265-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Skull surgery (with optional tracer/muscle implant/wireless device implant) 2. Spinal injury (with optional ChABC/cell injections)*, minimum of 1 week following #1 3. Spinal surgery* (electrodes/waveguide and ChABC/cells/virus/tracer) with optional muscle/wireless device implant minimum of 1 week following #2 4. Brain tracer injections*, minimum of 1 week following #3 5. Spinal Tracer Injections*, minimum of 1 week following #4 6. Superficial repair surgery*, minimum of 1 week following any of the above surgeries <p>* Some of these surgeries will be omitted for many of the animals</p> <p>Species: Rats Interval: Minimum 1 week Rationale: Multiple survival surgeries are required to create this complex animal model to test the combination of stimulation and differentiated cell therapies at restoring movement following spinal cord injury.</p> <ul style="list-style-type: none"> - The initial skull surgery must be separated from the remaining surgeries in order to determine the effects of brain recording and/or stimulation prior to injury of the spinal cord - When differentiated cells are injected, two spinal surgeries must be performed because differentiated cells cannot be implanted at the time of injury due to the pro-scarring and reactive astrocyte cues present during acute injury. - The brain tracer must be injected after recovery has taken place due to our experimental intervention. This is a minor, 20-minute procedure involving only a small bur hole through the skull given that the animal will already have an acrylic head cap placed during the skull surgery. Spinal tracers must often be injected in a separate surgery than brain tracers due to different propagation times. - During these long-term studies, it may be occasionally necessary to add sutures to the skin near the head implant to protect a catheter after the skin has relaxed or eroded. Head caps also become lose over time, and it may be necessary to add several skull screws and new acrylic in a surgical procedure. We will consult with veterinary services before performing such repairs. Note that we will not re-approach the spinal cord in these procedures. Repairs will only involve the skin, skull, and superficial muscle layers. <p>or</p> <ol style="list-style-type: none"> 1 Spinal or Cortical Viral Injections 30+ days 2 Spinal injury and waveguide/microLED implant 7+ days 3 Brain Tracer Injections* 7÷ days

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	<p>4 Spinal Tracer Injections* 7+ days 5 Repair surgery (if needed)* 7+ days 6 Terminal Procedure Electrophysiology and Perfusion* 1+ day Species: Rats Interval: 2-4 days Rationale: The brain tracer must be injected after recovery has taken place due to our experimental intervention. This is a minor, 20-minute procedure involving only a small bur hole through the skull given that the animal will already have an acrylic head cap placed during the skull surgery. Spinal tracers must often be injected in a separate surgery than brain tracers due to different propagation times. During these long-term studies, it may be occasionally necessary to add sutures to the skin near the head implant to protect a catheter after the skin has relaxed or eroded. Head caps also become loose over time, and it may be necessary to add several skull screws and new acrylic in a surgical procedure. We will consult with veterinary services before performing such repairs. Note that we will not re-approach the spinal cord in these procedures. Repairs will only involve the skin, skull, and superficial muscle layers. * Some of these surgeries will be omitted for many of the animals</p> <p>OR</p> <p>Multiple Major Surgery (V. 108) 1. Viral transfection, requiring bladder wall exposure 2. Peripheral neural interface implantation, at least 14 days after #1 Species: Rats Interval: At least 14 days Rationale: Multiple surgeries will be avoided if possible by implanting the peripheral neural interface at the same time as the viral transfection. However, transgene expression can take several weeks (up to 20) to reach peak levels following transfection, and the recording quality from the peripheral neural interface is expected to degrade over time, so it may be necessary to implant the neural interface during a second surgery to ensure both high levels of expression and high quality electrophysiological recordings.</p>
4266-04	<p>Multiple Surgeries Table 1) A) Endoscopic colonic biopsy 2) A) Endoscopic colonic biopsy 3) A) Endoscopic colonic biopsy B) Jejunal endoscopic biopsy 4) A) Endoscopic colonic biopsy B) Jejunal endoscopic biopsy 5) A) Endoscopic colonic biopsy B) Jejunal endoscopic biopsy 6) A) Endoscopic colonic biopsy B) Jejunal endoscopic biopsy 7) A) Endoscopic colonic biopsy B) Laparoscopic MLN 8) A) Peripheral (axillary or inguinal) lymph node biopsy B) Up to 15 cm transection the jejunum and associated mesenteric Lymph nodes (separated out) 9) A) Endoscopic colonic biopsy 10) A) Endoscopic colonic biopsy 11) A) Peripheral (axillary or inguinal) lymph node biopsy B) Up to 15 cm transection the jejunum and associated mesenteric Lymph nodes (separated out) 12) A) Peripheral (axillary or inguinal) lymph node biopsy B) Up to 15 cm transection the jejunum and associated mesenteric Lymph nodes (separated out) 13) A) Endoscopic colonic biopsy 14) A) Endoscopic colonic biopsy B) Laparoscopic MLN 15) A) Endoscopic colonic biopsy 16) A) Endoscopic colonic biopsy B) Laparoscopic MLN 17) A) Endoscopic colonic biopsy 18) A) Endoscopic colonic biopsy Time between Surgery 1 and 2: 1-2 weeks _ refer to note € below for clarification on timings, that applies to timing between all surgeries. Species: M. mulatta Interval >1- >6 weeks Rationale for multiple surgical sessions in a single animal: *Lymph nodes are where the initial immune cells interact with components of the vaccine. This system is also separate from the gut and blood. Since</p>

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	<p>populations of immune cells change during the course of the study, it is important that lymph nodes are repeatedly analyzed in order to determine vaccine induced immune responses. Mesenteric lymph nodes in particular, can be better predictors of vaccine efficacy. Measuring vaccine-induced immune responses in this tissue serves, as an important anatomical site for viral replication, will be a better predictor of vaccine efficacy.</p> <p>*Colon & Jejunal biopsies will be collected to provide additional time points to measure the impact of the vaccine on the viral reservoir. Since a maximum of only 5 million cells can be routinely isolated from this sample, it is adequate for analyzing viral replication but does not provide a sufficient number of cells to analyze T cell responses, as does the jejunal resection surgery. Samples collected from the biopsy will provide time points between the jejunal resections.</p> <p>*Jejunum and associated mesenteric lymph nodes will be collected to measure virus and immune responses in the gut. The time points selected will provide analysis of a baseline immunological and virological data, which must be established prior to vaccination, so that any alteration in the immune response can be attributed to vaccination. These sampling are essential to profile the immune and virological response to vaccine induced protection.</p>
4278-02	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Craniotomy - EEG, thermistor 2. CCI, osmotic pump, guide cannula implant, minimum 10 days following #1 <p>Species: Mice Interval: 10 days</p> <p>Rationale: Animals are used in a within-subject design to study sleep-wake behavior, EEG, and brain temperature both before and after controlled cortical impact (CCI). To obtain the control data the EEG and thermistor implants must be made before the CCI is conducted.</p>
4286-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Cardiac injury 2. Intramyocardial cell transplantation, 7 days following #1 <p>Species: Guinea Pigs and Rats Interval: 7 days</p> <p>Rationale: These procedures must be performed serially to ensure cell survival (graft survival would be expected to be reduced during the very acute phase following cardiac injury) and to model the expected situation in human patients (in which cells would likely be grafted into the heart in the healing phase of infarction, rather than immediately after coronary occlusion).</p>
4310-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Eye coil and Head lug implantation 2. Craniotomy and implantation of 1 to 3 recording cylinder/chamber, minimum of 2 weeks following #1. 3. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Species: Macaca mulatta Interval: At least 2 weeks</p> <p>Rationale: Multiple surgical sessions minimize craniotomy exposure time, repair surgeries maximize the use of animals (by repairing damaged implants), and, therefore, minimize the number of animal used in the project. We are requesting a repair surgery per the IACUC Guidelines for Non-human Primate Neuroscience Studies.</p>
4314-01	<p>ACTIVE Version 3 Bacterial vaginosis study, Aim I Aim I, Group I (control)</p> <p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection 2. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #4 6. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #5

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<p>7. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #6 8. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #7 9. Vagina, jejunum, colon, and rectum biopsy and CSF collection, at least 4 weeks after #8 10. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #9 Species: Macaques Interval: At least 1 week to at least 4 weeks Rationale: The importance of this study is to understand the kinetics of the mucosal and peripheral immune system in relation to changes in hormone levels and the microbiome. Multiple biopsies and colon/jejunum resection surgeries will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to differing treatments and phases of the menstrual cycle. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies and resection are our only options to collect immune cells for this study. Ten biopsy/CSF/resection time points were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV, during treatment prior to SIV, and after SIV infection in this control group.</p> <p>ACTIVE Version 3 continued Bacterial vaginosis study, Aim I, continued Aim I, Groups II-III (BV,BV+Probiotics) Multiple Major Surgery: 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection 2. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #4 6. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #5 7. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #6 8. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #7 9. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #8 10. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #9 11. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #10 Species: Macaques Interval: At least 1 week to at least 4 weeks Rationale: The importance of this study is to understand the kinetics of the mucosal and peripheral immune system in relation to changes in hormone levels and the microbiome relative to vaginosis, and to develop a model of bacterial vaginosis. Multiple biopsies and colon/jejunum resection surgeries will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to differing treatments and phases of the menstrual cycle. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies and resection are our only options to collect immune cells for this study. Eleven biopsy/CSF/resection time points were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV, during treatment prior to SIV, and after SIV infection in these BV and BV + probiotic groups</p> <p>ACTIVE Version 4 (Note: Version 4, Replaces Aim II timelines of version 3 (and modified in version 5) (Note: There is no multiple major surgery in this time line; included for improved understanding of timelines) Group A: Pre-SIV treatment *Multiple Surgery* 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection</p>
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<p>2. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #4 6. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #5 7. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #6 8. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #7 9. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #8 10. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #9 11. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #10 12. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #11 13. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #12 14. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #13 Species: Macaques Interval: At least 1 week Rationale: The importance of this study is to determine the effects of Lacto/IL-22 treatment on mucosal immunity and markers of mortality during SIV infection. Multiple biopsies and CSF collections will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to Lacto/IL-22 treatments, SIV infection, and phases of the menstrual cycle. Due to the nature of mucosal immunity and limited sampling techniques available, biopsies and CSF are our only options to collect immune cells for this study. Fourteen biopsy/CSF timepoints were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV (timepoint 1), during acute SIV infection (timepoints 2-6), and during chronic SIV infection and treatment (timepoints 7-14). Fourteen timepoints were selected in order to determine the kinetics of mucosal changes prior to SIV, acute infection, chronic infection and during treatment, and late/chronic disease progression. These time points are essential in order to be able to get enough cells for different assays and to effectively characterize each stage. Given the length of time throughout this study, the large number of time points is adequately spread out.</p> <p>ACTIVE Version 4 continued (Note: Version 4, Replaces Aim II timelines of version 3 (and modified in version 5) (Note: There is no multiple major surgery in this time line; included for improved understanding of timelines) Group B: Chronic/ART *Multiple Surgery*</p> <p>1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection 2. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 2 weeks after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #3 5. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #4 6. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #5 7. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #6 8. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #7 9. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #8 10. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #9 11. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #10 12. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #11 13. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #12 14. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #13 Species: Macaques Interval: At least 1 week Rationale: The importance of this study is to determine the effects of Lacto/IL-22 treatment on mucosal immunity and markers of mortality during SIV infection. Multiple biopsies and CSF collections will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to Lacto/IL-22 treatments, SIV infection, and phases of the menstrual cycle. Due to the nature of mucosal immunity and limited sampling techniques available, biopsies and CSF are our only options to</p>
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	<p>collect immune cells for this study. Fourteen biopsy/CSF timepoints were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV (timepoint 1), during acute SIV infection (timepoints 2-6), and during chronic SIV infection and treatment (timepoints 7-14). Fourteen timepoints were selected in order to determine the kinetics of mucosal changes prior to SIV, acute infection, chronic infection and during treatment, and late/chronic disease progression. These time points are essential in order to be able to get enough cells for different assays and to effectively characterize each stage. Given the length of time throughout this study, the large number of time points is adequately spread out.</p> <p>ACTIVE Version 14 (ammendment to version 12, FMT study) for animals in Group B,I, surgery schedule adds option for minor or major biopsy at each time point and omits vaginal biopsy since all males: Group B, I *Multiple Surgery*</p> <ol style="list-style-type: none"> 1. Lymph node, jejunum, colon and rectum (6 major or 24 minor pinch biopsies) biopsy 2. Lymph node, jejunum, colon and rectum (6 major or 24 minor pinch biopsies) biopsy, at least 1 week after #1 3. Lymph node, jejunum, colon and rectum (6 major or 24 minor pinch biopsies) biopsy, at least 1 week after #2 4. Lymph node, jejunum, colon and rectum (6 major or 24 minor pinch biopsies) biopsy, at least 1 week after #3 5. Lymph node, jejunum, colon and rectum (6 major or 24 minor pinch biopsies) biopsy at least 1 week after #4 <p>Species: Macaques Interval: At least 1 week Rationale: The importance of this study is to determine the effects of fecal transplant treatment on mucosal immunity and markers of mortality during SIV infection, with the goal to decrease the virus reservoir. Multiple biopsy collections will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to the transplant. Due to the nature of mucosal immunity and limited sampling techniques available, biopsies are our only options to collect immune cells for this study. Four time points were selected in order to determine the kinetics of mucosal changes prior to and early after treatment. These time points are essential in order to be able to get enough cells for different assays and to effectively characterize each stage. *Lymph nodes will be palpated and if none are felt, surgery will not be performed.</p> <p>ACTIVE Version 4 continued (Note: Version 4, Replaces Aim II timelines of version 3 (and modified in version 5) (Note: There is no multiple major surgery in this time line; included for improved understanding of timelines) Group C: Chronic/ART Treatment *Multiple Surgery*</p> <ol style="list-style-type: none"> 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection 2. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 2 weeks after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 2 weeks after #4 6. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #5 7. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #6 8. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #7 9. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 2 weeks after #8 10. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #9 11. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #10 12. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #11 13. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #12
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<p>14. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #13 15. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #14 16. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #15 Species: Macaques Interval: At least 1 week Rationale: The importance of this study is to determine the effects of Lacto/IL-22 treatment on mucosal immunity and markers of mortality during SIV infection. Multiple biopsies and CSF collections will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to Lacto/IL-22 treatments, SIV infection, and phases of the menstrual cycle. Due to the nature of mucosal immunity and limited sampling techniques available, biopsies and CSF are our only options to collect immune cells for this study. Sixteen biopsy/CSF timepoints were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV (timepoint 1), during acute SIV infection (timepoints 2-3), then if UNINFECTED, another pre-SIV timepoint (surgery 4), re-infect SIV then acute SIV #2 (surgeries 5-8), and during chronic SIV infection and treatment (timepoints 9-16). Sixteen timepoints were selected in order to determine the kinetics of mucosal changes prior to SIV, re-infection if not infected, acute infection, chronic infection and during treatment, and late/chronic disease progression. These time points are essential in order to be able to get enough cells for different assays and to effectively characterize each stage. Given the length of time throughout this study, the large number of time points is adequately spread out.</p> <p>ACTIVE Version 9 Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Colon, rectal and lymph node biopsy 2. Colon, rectal and lymph node biopsy, at least 1 week after #1 3. Colon, rectal and lymph node biopsy, at least 1 week after #2 4. Colon and rectal biopsy, at least 1 week after #3 5. Colon, rectal and lymph node biopsy, at least 1 week after #4 6. Colon, rectal and lymph node biopsy, at least 1 week after #5 7. Colon and rectal biopsy, at least 1 week after #6 8. Colon, rectal and lymph node biopsy, at least 1 week after #7 <p>Species: Macaques Interval: At least 1 week Rationale: The importance of this study is to understand the kinetics of vaccine-specific immune responses in mucosal tissues. Multiple biopsies will be essential to determine the kinetics of the mucosal and lymphoid immune response prior to and after vaccination. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies are our only options to collect immune cells for this study. Surgery 8 may only occur if long-term follow up is deemed necessary as determined by high levels of vaccine responses that may confer resistance for an extended duration.</p> <p>INACTIVE Note version 4 replaces version 3 time line: Version 3, Aim II, Group I (Acute SIV) (see version 5 below for revised surgery plan) Multiple Major Surgery</p> <ol style="list-style-type: none"> 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection 2. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #1 3. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #2 4. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #4 6. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #5 <p>Interval: At least 1 week to at least 4 weeks Rationale: The importance of this study is to understand the kinetics of the mucosal and peripheral immune system in relation to changes in hormone levels and the microbiome. Multiple biopsies and</p>

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<p>colon/jejunum resection surgeries will be essential to determine the kinetics of the mucosal and lymphoid immune response over time and in response to differing treatments and phases of the menstrual cycle. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies and resection are our only options to collect immune cells for this study. Ten biopsy/CSF/resection time points were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to treatment and SIV, during treatment prior to SIV, and after SIV infection in this control group.</p> <p>INACTIVE Note version 4 replaces version 5 time line: Version 5, for Aim II, Group I, clarifies re-challenge/re-cycle through acute phase a maximum of 3 times and removes CSF collection and colon/jejunum resection Aim II, Group I (Acute SIV) Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Lymph node, vagina, jejunum, colon and rectum biopsy 2. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 1 week after #1 3. Vagina, jejunum, colon and rectum biopsy, at least 1 week after #2 4. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 1 week after #3 <p>*if not infected, continues below; if infected move to chronic phase*</p> <ol style="list-style-type: none"> 5. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 3 weeks after #4 6. Vagina, jejunum, colon and rectum biopsy, at least 1 week after #5 7. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 1 week after #6 <p>*if not infected, continues below; if infected move to chronic phase*</p> <ol style="list-style-type: none"> 8. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 3 weeks after #7 9. Vagina, jejunum, colon and rectum biopsy, at least 1 week after #8 10. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 1 week after #9 <p>*if not infected, continues below; if infected move to chronic phase*</p> <ol style="list-style-type: none"> 11. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 3 weeks after #10 12. Vagina, jejunum, colon and rectum biopsy, at least 1 week after #11 13. Lymph node, vagina, jejunum, colon and rectum biopsy, at least 1 week after #12 <p>Species: Macaques Interval: At least 1 week to at least 4 weeks Rationale: The importance of this study is to understand the kinetics of the mucosal and peripheral immune system during acute SIV infection. Multiple biopsies will be essential to determine the kinetics of the mucosal and lymphoid immune response over time prior to and after SIV infection. Animals will be given a 30% infectious SIV dose in order to investigate responses in infected versus exposed, uninfected to SIV challenge. Any animals that are not infected will be moved back to challenge phase, up to 3 times (maximum of 4 challenges total). On the 4th challenge, a high dose (100% infectious dose) SIV challenge will be given to animals to ensure infection. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies are our only options to collect immune cells for this study. Up to 13 biopsy time points were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to and after SIV infection, in this acute SIV infection group.</p> <p>INACTIVE Note version 4 replaces version 3 timeline: Version 3 continued, Aim II, Group II (Chronic SIV) Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection

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	<p>2. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #1 3. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #2 4. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #3 5. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #4 6. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #5 7. Colon and jejunum resection, lymph node, vagina, and rectum biopsy, and CSF collection, at least 2 weeks after #6 8. Vagina, jejunum, colon and rectum biopsy and CSF collection, at least 4 weeks after #7 9. Lymph node, vagina, jejunum, colon and rectum biopsy and CSF collection, at least 1 week after #8 Species: Macaques Interval: At least 1 week to at least 4 weeks Rationale: The importance of this study is to understand the kinetics of the mucosal and peripheral immune system during chronic SIV infection. Multiple biopsies and colon/jejunum resection surgeries will be essential to determine the kinetics of the mucosal and lymphoid immune response over time prior to and after SIV infection. Due to the nature of mucosal immunity and the limited sampling techniques available, biopsies and resection are our only options to collect immune cells for this study. Six biopsy/CSF/resection time points were selected in order to perform the needed experiments to assess mucosal immune changes with limited cell numbers isolated from biopsies in a longitudinal manner prior to and after SIV infection, in this chronic SIV infection group.</p> <p>Version 15, Rechallenge: For updated multiple surgery table see the significant change - makes changes to version 3, 4 and 5.</p>
4315-01	<p>Multiple Major Surgery: 1. Lymph node biopsy 2. Lymph node biopsy, at least 1 week following #1 3. Implantation of indwelling catheter(1 or 2 ver 2), at least 1 week following #2 4. Repair of indwelling catheter, at least 1 day following #3 5. Lymph node biopsy, at least 1 week following #4 6. Repair of indwelling catheter, at least 1 day following #5 7. Nephrectomy, at least 1 day following #6 8. Renal Transplant including removal of the second native kidney, at least 2 weeks following #7 9. Lymph node biopsy, at least 1 week after #5 and at least 30 days following #8</p> <p>Species: M. Mulatta Interval: At least 1 day to at least 30 days Rationale: These multiple surgeries are required for the longitudinal analysis of the impact of NK depletion (Group 1), costimulation blockade (Group 3) and Tregs (Group 4) on immune tolerance induction. The lymph node biopsies allow us to analyze the impact of the experimental protocol on the primary lymph organs, to determine if the immunologic response that we measure in the peripheral blood is also occurring in the sites of antigen presentation (the lymph nodes). The indwelling catheter placement allows us to provide optimal clinical supportive care for the transplant recipients. Given that these catheters are expected to stay in place for as long as 100 days, we have included two potential repairs in our multiple surgery plan, which may be necessitated for a mechanical problem. We are also including the possibility of reimplantation of the catheter line even after prior catheter removal in case of clinical necessity. Finally, renal transplantation is performed to most rigorously test whether immune tolerance is induced by our experimental protocol. Recipients first undergo a single nephrectomy and then, at least two weeks later, a renal transplant and concomitant removal of the second native kidney.</p> <p>Groups 1,2a,2b, 3, 4 Using animals that were previously used as leukopheresis donors as transplant recipients. Multiple Major Surgery: 1. Surgical approach to the femoral vein for placement of apheresis catheter 2. Surgical approach to the femoral vein for placement of apheresis catheter, at least 4 weeks after #1 3. Lymph node biopsy, at least 4 weeks after #2</p>

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	<p>4. Lymph node biopsy, at least 1 week after #3 5. Implantation of Indwelling Catheter (1 or 2, ver 2), at least 1 week after #4 6. Repair of Indwelling Catheter, at least 1 day after #5 7. Lymph node biopsy, at least 1 week after #6 8. Lymph node biopsy, at least 1 week after #7 9. Lymph node biopsy, at least 1 week after #8 10. Repair of Indwelling Catheter, at least 1 day after #9 11. Lymph node biopsy, at least 1 week after #10 12. Lymph node biopsy, at least 1 week after #11 13. Nephrectomy, at least 1 week after #12 14. Renal Transplant including removal of the second native kidney, at least 2 weeks after #9 15. Lymph Node Biopsy, at least one week after the last lymph node biopsy and at least 30 days after renal transplantation. 16. Re-implantation of Vmed Device, will be performed as needed and not sooner than 1 day following any other procedure.</p> <p>Species: M. Mulatta Interval: At least 1 day to at least 30 days Rationale: These multiple surgeries are required for the longitudinal analysis of the impact of NK depletion (Group 1) CAR+ T cells (Group 2b), costimulation blockade (Group 3) and Tregs (Group 4) on immune tolerance induction in animals who have previously been utilized as peripheral blood stem cell donors. Utilizing previous leukopheresis donors in this fashion is necessary because transplant pairs are chosen based on rare MHC compatibility criteria. Thus, there are occasions where animals that had previously been donors are found to have these rare MHC compatibilities and must be used. The lymph node biopsies allow us to analyze the impact of the experimental protocol on the primary lymph organs, to determine if the immunologic response that we measure in the peripheral blood is also occurring in the sites of antigen presentation (the lymph nodes). The indwelling catheter placement allows us to provide optimal clinical supportive care for the transplant recipients. Given that these catheters are expected to stay in place for as long they are functional, we have included two potential repairs in our multiple surgery plan, which may be necessitated for a mechanical problem. We are also including the possibility of reimplantation of the catheter line even after prior catheter removal in case of clinical necessity. Finally, renal transplantation is performed to most rigorously test whether immune tolerance is induced by our experimental protocol. Recipients first undergo a single nephrectomy and then, at least two weeks later, a renal transplant and concomitant removal of the second native kidney.</p>
4316-01	<p>Multiple Major Surgery: Standard approach. 1. Head post implant 2. Chamber placement >8 weeks after #1 3. Craniotomy > 4 weeks after #2 4. Lesion (6 animals only) >1 year after #3 5. Second chamber surgery (if needed) Estimated >6 mo after #4 6. Repair/correction surgery Species: M. mulatta Interval: See above. Rationale: Previous surgical experience where head post, chamber placement, and craniotomy were done in fewer surgeries resulted in less stable implants and increased discomfort and prolonged healing time for the animals. Training on eye fixation tasks following a head post surgery will be necessary and delaying the chamber and craniotomy will minimize the risk of infection. Introduction of the ibotenate will all sufficient neural recordings before the lesion. A second chamber surgery may be necessary to complete the aims of the scientific study if additional neural recordings are needed in the other hemisphere or for comparison of neural activity between hemispheres in a single monkey. One repair surgery is requested per animal if unforeseen repair, removal or replacement of an implant is required. Repairs will be considered whenever they would not jeopardize the health of the animal.</p> <p>Version #14 Multiple Major Surgery: Flexible arrays reduce recovery time. Recording, no lesion:</p>

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	<ol style="list-style-type: none"> 1. Head post implant 2. Chamber placement >2 weeks (if already eye-fixation trained) or >8 weeks after #1 3. Craniotomy and/or chronic implant placement > 1 week after #2 4. Craniotomy and/or chronic implant placement > 1 month after #3 5. Second chamber surgery (if needed) > 1 month after #4 6. Craniotomy and/or chronic implant placement > 1 week after #5 7. Craniotomy and/or chronic implant placement > 1 month after #6 8. Repair/correction surgery > 1 month after any other surgery. <p>Behavior and lesion, no recording:</p> <ol style="list-style-type: none"> 1. Head post implant 2. Lesion > 1 month after #1 3. Repair/correction surgery > 1 month after any other surgery. <p>Recording and lesion:</p> <ol style="list-style-type: none"> 1. Head post implant 2. Chamber placement >2 weeks (if already eye-fixation trained) or >8 weeks after #1 3. Craniotomy and/or chronic implant placement > 1 week after #2 4. Second chamber surgery (if needed) > 1 month after #3 5. Craniotomy and/or chronic implant placement > 1 week after #4 6. Lesion > 1 year after #5 we cannot do any recording experiments once the lesion has been performed on the target structures 7. Repair/correction surgery > 1 month after any other surgery. <p>Behavioral control, no lesion:</p> <ol style="list-style-type: none"> 1. Head post implant 2. Chamber placement >2 weeks (if already eye-fixation trained) or >8 weeks after #1 3. Craniotomy and/or chronic implant placement > 1 week after #2 4. Second chamber surgery (if needed) > 1 month after #3 5. Craniotomy and/or chronic implant placement > 1 week after #4 6. Repair/correction surgery > 1 month after any other surgery. <p>Species: <i>M. mulatta</i> Interval: See above. Rationale: Surgical implantation of the headpost and recording chamber and creation of the craniotomy or implantation of flexible arrays are all necessary to successfully record neural activity in awake, behaving monkeys. Previous surgical experience where these procedures were combined into fewer surgeries have resulted in less stable implants and increased discomfort and prolonged healing time for the animals. However, with the procedure described in this amendment which includes covering small burr holes and reducing the need for maintenance of an exposed craniotomy, the expected discomfort and healing time between surgery 2 and 3 and 5 and 6 can be significantly decreased. Additionally, training on eye fixation tasks following a headpost surgery will be necessary and delaying the chamber and craniotomy until after this training is complete will minimize the risk of infection. In the event that an animal is already trained on eye fixation tasks (as in the case of a replacement or repair of the headpost implant on a previously trained animal), a subsequent surgery may be scheduled sooner although the time allowed for total healing of the headpost before use will not be shortened from our approved protocol (4-6 weeks from head post implant surgery to use of the head post for head stabilization during testing). The advantage to placing these procedures closer together allows immediate data collection once arrays are implanted, maximizing the usable data collection time and potentially minimizing the number of repeated implant placement procedures overall. A second chamber surgery will follow the same rationale as in our existing protocol, but placement of a second chamber may be determined by the ability to access the desired structures with additional flexible array implantations in the original hemisphere as well as quality of neural signals and total number of penetrations in the original chamber.</p>
4321-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Intracranial cannulation osmotic pump implant 2. Osmotic pump explant 19-25 days later

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	<p>Species: Mice Interval: 19-25 days Rationale: Need to explant the pump to obtain precise offset of drug administration.</p> <p>Multiple Major Surgery: 1. Injection surgery 2. Intracranial cannulation 19-25 days later Species: Mice Interval: 19-25 days Rationale: The injection surgery is necessary to introduce expression of light-sensitive channels to specific brain regions. These channels then respond to optic stimulation, which will be delivered by insertion of a fiber optic cable through the second surgery. The cannulation cannot be made at the same time because the implant would degrade by the time the channels have become expressed.</p>
4324-02	<p>Multiple Major Surgery: 1. Preconditioning kidney ischemia and reperfusion (Bilateral) 2. Ischemia-reperfusion of the kidney (Bilateral) Species: Mice Interval: 2-14 days Rationale: Preconditioning allows efficient kidney injury.</p> <p>OR</p> <p>Multiple Major Surgery: 1. Virus injection into kidney (Bilateral) WITHOUT preconditioning kidney ischemia and reperfusion 2. Ischemia-reperfusion of the kidney (Bilateral) Species: Mice Interval: 2-14 days Rationale: Viral vectors require at least 1-2 days before transgenes are expressed at a sufficient level.</p> <p>OR (v. 4)</p> <p>Multiple Major Surgery: 1. Virus injection into kidney 2. Unilateral ureteral obstruction (UUO), 2-14 days after #1 Species: Mice Interval: 2-14 days Rationale: Virus injection into the kidney will allow genes to be expressed before UUO for our studies to identify genetic factors regulating kidney fibrosis.</p>
4348-01	<p>Multiple Major Surgery: 1. Recording chamber (up to 3) and titanium restraining hardware implant, craniotomy and maybe scleral search coil implant 2. Scleral search coil implant in second eye, or both if not done in 1, minimum of 2 weeks following #1. 3. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. Species: M. Mulatta Interval: At least 2 weeks Rationale: We need to implant single unit recording chambers over oculomotor regions of the brain to permit neurophysiological studies. We usually perform the surgery consisting of 1 coil implant, chambers and head stabilization post all in one surgery. Even an experienced team needs 4-6 hours to accomplish all of this. Therefore, we perform a second scleral search coil surgery (at least 2 weeks after the first), to reduce the trauma to the monkey. However, if surgery #1 takes longer than usual, we may elect to implant coils in both eyes during surgery #2. If no repairs are needed, group I animals will undergo no more than 2 surgeries. Our scleral search coil implants are minimally disruptive to the animal. Placing the second coil two weeks or more after the first allows the monkey to recover optimally. There is little to no swelling of orbital tissues in most cases. A broken coil might need to be replaced. A coil may break due to defective</p>

Appendix C – Multiple Major Surgeries

	<p>insulation or metal fatigue.</p> <p>Group II, Infants Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Eye muscle surgery 2. Recording chamber (up to 3) and titanium restraining hardware implant, craniotomy and maybe scleral search coil implant. 3. Scleral search coil implant in second eye, or both if not done in 1, minimum of 2 weeks following #2. 4. Potential repair surgery, as described in the approved UW IACUC Guidelines for Non-human Primate Neuroscience Studies, interval depends on need, but is always preceded by a veterinary pre-surgical assessment. <p>Species: M. Mulatta Interval: 1.5 - 2 years between surgery #1 and #2; At least 2 weeks between #2 and #3 Rationale: We have developed effective animal models for strabismus using surgical methods like those used in human infants. To achieve our goals, surgical adjustment of eye muscles must be made during infancy (critical period for visual-oculomotor development). When surgical animals reach at least 1.5 - 2 years of age, they will be subjects in studies of neural control of eye movements as described under our juvenile protocols. This is where the 2nd and 3rd surgical surgical procedures are necessary. We need to implant single unit recording chambers over oculomotor regions of the brain to permit neurophysiological studies. We usually perform the surgery consisting of coil implant, chambers and head stabilization post all in one surgery. Even an experienced team needs 4-6 hours to accomplish all of this. Therefore, we perform a second scleral search coil surgery (at least 2 weeks after the first), to reduce the trauma to the monkey. However, if surgery #2 takes longer than usual, we may elect to implant coils in both eyes during surgery #3. If no repairs are needed, group II animals will undergo no more than 3 surgeries. Our scleral search coil implants are minimally disruptive to the animal. Placing the second coil two weeks or more after the first allows the monkey to recover optimally. There is little to no swelling of orbital tissues in most cases. A broken coil might need to be replaced. A coil may break due to defective insulation or metal fatigue.</p>
4349-02	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Creating rabbit liver VX2 tumor 2. Drug Eluting Bead Chemoembolization (DEB-TACE) 3. Imaging-guided percutaneous radiofrequency ablation (RFA) <p>Species: Rabbit Interval: 15-21 days between 1 and 2 Interval: 3-5 days between 2 and 3 Rationale: First surgery for tumor model creation Second and third surgeries for tumor DEB-TACE and RFA sequential treatment procedures.</p> <p>Or</p> <p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Creating rabbit liver VX2 tumor 2. Imaging-guided percutaneous radiofrequency ablation (RFA) 3. Drug Eluting Bead Chemoembolization (DEB-TACE) <p>Species: Rabbit Interval: 15-21 days between 1 and 2 Interval: 3-5 days between 2 and 3 Rationale: First surgery for tumor model creation Second and third surgeries for tumor RFA and DEB-TRACE.</p>
4356-01	<p>Multiple Major Surgery:</p> <ol style="list-style-type: none"> 1. Intracranial AAV injection 2. Cranial window, head plate, and nasal cannula implantation, >2 weeks after #1 <p>Species: Mice Interval: Greater than 2 weeks Rationale: The two surgeries must happen separately because the virus requires at least 2 weeks to express the light-activated protein at adequate levels for visualization and the length of time that the cranial window will remain clear is uncertain, so in order to</p>

Appendix C – Multiple Major Surgeries

	<p>reliably obtain data implantation of the cranial window must wait until the virus has reached usable levels of expression. The head plate and nasal cannula implants will be installed in the same session as the cranial window.</p>
4362-01	<p>Multiple Major Surgery: 1. Laminectomy/Spinal Cord Contusion 2. Spinal Decompression (aim 1 & 2), approximately 2 days following #1 3. Tracer injection (aim 1 & 3), 10-11 weeks posts injury. Species: Rat Interval: 2 days between surgeries 1 & 2 and 10-11 weeks between surgeries 3 & 4. Rationale: For Aim 1, the main purpose is to determine the effects of spinal decompression and its effects on the secondary damage caused by the swelling after injury. Therefore, it is necessary to perform this second surgery to decompress the swollen spinal cord. A third surgery (tracer injections) at 10-11 weeks post injury is also proposed to trace the regenerating axons. For Aim 3, it is necessary to determine the extent and origin of the regenerated axons so that can accurately determine the effects of the scaffolds on axonal regeneration.</p>
4370-01	<p>Multiple Major Surgery: 1. Lami/Spinal Contusion 2. Craniotomy/Cortical Injection and/or Intraspinal Injection with Wave guide/Ferrule implant* (Injections with differentiated virally transfected cells or virus). 3. Tracer Injection (cortical and/or spinal cord) (BDA, FluoroRuby, FastBlue, Microspheres, Fluorogold)* 4. Intramuscular Tract Tracer Injection (PRV)* 5. Repair Surgeries In case a repair to skin, skull, and superficial muscle layers is required we will contact Veterinary Services for their advice about whether it is clinically appropriate to proceed, and with their concurrence and advice we will attempt to reattach the ferrule. If erosion of the spinal implants occurs the animal will be sacrificed. The lab's standard SOP for spinal injuries (attached) will be followed. Species: Rat Interval between 1 and 2: 10-14 Days Interval between 2 and 3: 2+ weeks Interval between 3 and 4: 1-2 weeks Interval between 4 and 5: Varies, see above. Rationale: When differentiated cells are injected, two spinal surgeries must be performed because differentiated cells cannot be implanted at the time of injury due to the pro-scarring and reactive astrocyte cues present during acute injury. The brain and spinal cord tracers must be injected after recovery has taken place due to our experimental intervention. This is a 20-minute procedure involving only a small bur hole through the skull given that the animal will already have an acrylic head cap placed during the skull surgery. During these long-term studies, it may be occasionally necessary to add sutures to the skin near the head implant to protect a catheter after the skin has relaxed or eroded. Head caps also become loose over time, and it may be necessary to add several skull screws and new acrylic in a surgical procedure. We will consult with veterinary services before performing such repairs. Note that we will not re-approach the spinal cord in these procedures. Repairs will only involve the skin, skull, and superficial muscle layers. * Some of these surgeries will be omitted for some animals.</p> <p>OR</p> <p>1. Lami/Spinal Contusion 2. Window Chamber Implantation Spinal Injection or Craniotomy/Red Nucleus Injection 3. Spinal Injection Imaging Session #1 4. Spinal Injection Imaging Session #2 5. Spinal Injection Imaging Session #3 6. Spinal Injection Imaging Session #4 7. Spinal Injection Imaging Session #5 Species: Mouse Interval between 1 and 2: 5-10 days Interval between 2 and 3: 11-16 days for 1st imaging session Interval between 3 and 4: 3-7 days</p>

Appendix C – Multiple Major Surgeries

	<p>Interval between 4 and 5: 3-7 days Interval between 5 and 6: 3-7 days Interval between 6 and 7: 3-7 days Rationale: For this study, it is critical to implant the window chamber at 5-10 days post-injury to measure properties of myelinating cells acutely as they begin the process of repair and regeneration and to allow us to ultimately establish the process of internodal plasticity. The multiple imaging sessions are required because the process of myelin repair following injury occurs over many weeks.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Postnatal spinal injection of NpCre virus 2. Laminectomy/contusion spinal cord injury 8-12 Weeks <p>Species: Mouse pup Interval between 1 and 2: 8-12 Weeks Rationale: For this study, it is critical to inject the NpCre virus at P2-5 to catch myelinating cells before they mature. The contusion spinal cord injuries will occur in these animals only once they have reached adulthood (aged 8-12 weeks) as this is consistent with our model of SCI in mice. These two surgeries will enable us to measure the properties of mature myelin acutely after an injury.</p> <p>OR</p> <ol style="list-style-type: none"> 1. Laminectomy/Spinal Cord Contusion 2. Spinal Injection <p>Species: Mouse Interval between 1 and 2: 5 to 7 days Rationale: To optimally capture development of myelin repair after injury, a minimum of 5 days after injury is required before the lentivirus should be injected into the injured spinal cord. In other experiments, the injection will include wnt3a-secreting fibroblasts to accelerate repair.</p> <p>OR (v. 149)</p> <ol style="list-style-type: none"> 1. Laminectomy/Spinal Cord Contusion 2. Spinal Injection 3. Cranial Pedestal and Electrode Implantation <p>Species: Mouse Interval between 1 and 2: 1 day Interval between 2 and 3: 7-8 days Rationale: The retrovirus injection procedure requires some "settling" of the injury but cycling progenitor cells (along with scar-forming astrocytes) begin to ramp up production in the first 24 hours so it is necessary to incorporate the retrovirus prior to differentiation within this window. A separate surgery from the contusion injury is also required to optimize the viability of the electrode apparatus during stimulation. Positional disruption of the implanted intramuscular lead wires is less likely when the second surgery can be conducted 1-2 weeks after injury. During these long-term studies, it may be occasionally necessary to add sutures to the skin near the head implant to protect a wire after the skin has relaxed or eroded. Head caps also become loose over time, and it may be necessary to add a skull screw and/or new acrylic in a surgical procedure. We will consult with veterinary services before performing such repairs. Note that we will not re-approach the spinal cord in these procedures. Repairs will only involve the skin, skull, and superficial muscle layers.</p>
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Protocol Number	Wire Bottom Cages
2070-06	Wire Bottom Cages: Species: Mice Rationale: Needed to separate feces from urine for urine flow measurements.
3104-01	Wire Bottom Cages: Species: Mice Rationale: For the last 48 hours of the in vivo reverse cholesterol transport experiment, mice will be housed individually in wire-bottom cages for collection and analysis of feces from each mouse.
3298-02	Wire Bottom Cages: Species: Mice Rationale: The metabolic cages are wire bottom. This is necessary for the measurements being made.
2359-04	Wire Bottom Cages: Species: Mice Rationale: Metabolic cages with wire floor and without bedding are used for a period of 1 - 5 days because urine is being collected continuously to measure the secretion of a radiolabeled version of a protein that is secreted in the urine. And the urine may be radioactive.
2224-05	Wire Bottom Cages: Species: Mice Rationale: Metabolic cages are necessary to keep urine separate from feces during a 24-hour collection of urine samples from uremic mice.
3437-01	Wire Bottom Cages: Species: Mice Rationale: Metabolism studies, collection of individual data.
4281-01	Wire Bottom Cages: Species: Mice Rationale: Once ticks have been attached to their backs, mice will be individually housed in static cages with wire floor inserts above a thin layer of sterilized water. This housing arrangement is necessary to prevent other mice from chewing or rubbing off the attached ticks, and to provide a means for catching engorged ticks when they fall off of mice subsequent to feeding. Mice will be housed in this fashion for 3-5 days while ticks feed. Once all ticks have fallen off, mice will be euthanized.
2183-02	Wire Bottom Cages: Species: Mice Rationale: Mice will be placed in a wire bottom cage as opposed to a cage with normal bedding. This apparatus, used to study feeding behavior, works by recognizing a completed circuit between the slightly electrified floor and the metal spouts at the front of the cage. The mouse provides the completion of the circuit while engaging in licking behaviors, enabling a computer to record the data. The addition of bedding to the cage could insulate the floor, not allowing a circuit to be completed.
4196-01	Wire Bottom Cages: Species: Mice Rationale: Nalgene metabolic cages with wire floor and without bedding are used for up to 10 days to allow for urine collection during study of water homeostasis.
2968-04	Wire Bottom Cages: Species: Mice Rationale: Metabolic cages are needed for urine collection. Regular cages with bedding would make accurate urine collection difficult because the bedding would absorb the urine.
2011-21	Wire bottom cages: Using metabolic cages for 16-hour mouse urine collection sessions.
4237-01	Wire Bottom Cages: Species: Mice Rationale: Mice will be housed individually in wire bottom cages for the duration(48 hours) of the experiment "Role of PLTP and CLU in HDL composition and function, in vivo sterol efflux assay" to allow feces collection and measurement of radioactivity.
Protocol Number	Weaning Variance

4104-01	<p>Weaning Variance: Wean at 28 days. Breeding trio (1 male + 2 females) plus up to 1 litter of pups up to 28 days of age. Pregnant females will be moved from the breeding cage to their own cage.</p> <p>Species: Mice, NODscid IL2-R-gamma chain knockouts.</p> <p>Rationale: These mice are immunodeficient and immature at 21 days of age. We propose to keep one adult female with her litter for up to 28 days of age.</p>
3123-01	<p>Weaning Variance: Transgenics on C57BL/6 background (C57BL/6, Hes5-GFP, PLP-CreER, Dcx-CreER, Rosa-YFP) to be weaned at 28 days of age. Using a trio breeding scheme, if a newborn litter is born, the older litter will be separated with their mother until weaned.</p> <p>Species: Mice</p> <p>Rationale: Because this is an inbred strain of mice, transgenics born on the C57BL/6 background tend to be smaller than pups born on other backgrounds and the litters from these strains are generally smaller and less frequent and the mice will be more robust if left to grow longer.</p>
3333-01	<p>Weaning Variance: Up to 28 days. Maximum number of adult mice in a cage is 5 with one litter counting as 1 adult mouse.</p> <p>Species: All MDX (dystrophic) mice.</p> <p>Rationale: Dystrophic mice have smaller litter sizes and less robust weanlings than the wild type controls. The lab has implemented this weaning variance for the health of the mice because there was a fair amount of mortality when the dystrophic mice were weaned at 21 days. While most of the colony (mdx) mice will be weaned between 21 to 24 days, the more fragile strains (dystrophin:utrophin double knockout [mdx:utrn^{-/-}; or DKO]) requiring a longer time with the parents and will be evaluated on a litter by litter basis. The stronger/larger members of the same litter will be weaned as early as possible. The double knockout mice are the most fragile and breeding cages will be set up with one male and one female to help prevent overcrowding. With careful vigilance, the intent is to reduce both weaning and neonatal mortality.</p>
4134-01	<p>Weaning Variance: Wean at 28 days of age</p> <p>Species: Mice, CHF1/Hey2 KO</p> <p>Rationale: These mice are generally small at 3 weeks and have decreased survival when weaned at this age. Better success is achieved when weaned up to 28 days postpartum.</p>
4231-01	<p>Weaning Variance: Wean at 28 days instead of 21 days</p> <p>Species: Mice, Id1^{+/-}-Id3^{-/-}</p> <p>Rationale: Because of their transcriptional defects, growth rate of Id1^{+/-}-Id3^{-/-} pups is also slower so that at 21 days their body size resembles that of a 15-16 day old wild type pup. Weaning of such pups clearly smaller in body size is delayed from 21 to 28 days post-birth. Because these knock-out mice are usually represented at low frequency within a litter (i.e., 1 out of 8-12 pups), it is expected that no more than 1 or 2 pups per breeder cage may be left for weaning at 28 days at any given time.</p>
4339-01	<p>Weaning Variance: Wean at 28 days, maximum in cage 1 male and 2 female adults with 1 litter.</p> <p>Species: CAR-null mice</p> <p>Rationale: This mouse strain has lower body weight compared to age-matched wild type mice.</p>
4321-01	<p>Weaning Variance: Wean no later than 25 days onto breeder chow 5058.</p> <p>Species: Mice, line Th fs/fs</p> <p>Rationale: Homozygotes Th fs/fs mice are adipic, aphagic, hypoactive, small, and need to be left with the mothers a little longer than normal. If delayed weaning overlaps with the birth of a new litter, then we will wean remove all but the homozygous mutants from the cage. Due to the expected genotype frequency of 1:22 for Th fs/fs mice we expect no more than 1 or 2 homozygous mice with extended weaning would remain with the new litter.</p>
4321-02	<p>Weaning Variance: Wean by 30 days of age. In General mice are weaned around 25 days when they look large enough to survive without mother. Some latitude works well for the mice. We have 2 females and 1 male in a breeding cage. We remove the older pups as soon as possible if the other dam delivers. If additional litters are born i.e. if a second litter is born to a dam prior to the older pups being weaned then the older pups are weaned at that time.</p> <p>Species: Mice, D1 & D2-GFP heterozygotic BAC-transgenics</p> <p>Rationale: Mice are underweight at 21 days wean mice at 30 days instead of 21 days for health of pups, based on recommendations from Jackson labs.</p>
2878-01	<p>Weaning Variance: Wean Min mutation mice at 26 days of age. These mice will be in trio breeding because they have small litter sizes, small pups and poor breeding efficiency as described in the IACUC policy regarding the number of mice maintained in breeding cages. The maximum number of mice to be</p>

	<p>allowed in a cage is 11 pups and 3 adults. If a new litter is born in the cage prior to weaning of the old litter (pups greater than 19 days of age) the old litters will be weaned or separated immediately.</p> <p>Species: Mice, Min Mutation, C57BL/6J-APC^{min}</p> <p>Rationale: These mice tend to be much smaller than the usual pups and are physically underdeveloped at 21 days and it is better for them to stay with the dam up to 25 days in order to grow to full size before being weaned.</p>
3373-01	<p>Weaning Variance: Wean at 26 to 28 days of age. Trio breeding is used and the litter size varies from 3-6 pups per litter. The group will insure that if a new litter is born in the cage prior to weaning of the old litter (pups greater than 21 days of age), the old litter will be weaned or separated immediately. These mice are closely monitored at least once a week and sometime more frequently.</p> <p>Species: Mice, phox, iNOS and iNOS-phox mice</p> <p>Rationale: We breed phox, iNOS, and phox-iNOS double mutant mice. These mice, phox and iNOS-phox mice, are deficient in the ability to generate a respiratory burst in phagocytic cells. Mice have to be on prophylactic sulfamethoxazole-trimethoprim sulfate in their drinking water to prevent spontaneous infection from their normal flora. These strains are generally too small at 21 days. We have found that an additional 5-7 days is sufficient for them to grow and be weaned successfully. The iNOS mice are much more robust, but the pups still tend to grow slower even though they are not on antibiotics.</p>
2348-01	<p>Weaning Variance: Wean at 28 days of age.</p> <p>Species: Mice, TCR transgenic, RAG null, TCRα null, TCRβg null, plt and Bim null</p> <p>Rationale: These lines develop more slowly than standard strains and at 21 days are too small and weak to self-feed. Will have (a) 1 adult male, 2 adult females, and 2 litters up to 21 days of age, or (b) 1 adult female, 1 litter up to 28 days of age and 1 litter under 21 days of age. May need to separate out a female and pups into a separate cage until the pups are weaned, e.g., if one female has two litters and the other female has one litter. In rare cases in which 13 or more pups 14 days of age or older exist in a cage of breeders, we will separate out one female and her pups to reduce the total number of mice per cage to an acceptable level.</p>
3298-02	<p>Weaning Variance: Wean at 28 days. Breeding cages will contain 1 male, 2 females and potentially 2 litters of mice.</p> <p>Species: Mice, alpha syntrophin KO</p> <p>Rationale: These mice have slower muscle development than normal and are therefore smaller than normal mice at 3 weeks.</p>
2343-01	<p>Weaning Variance: Wean at 22-28 days of age.</p> <p>Species: Mice, PON1^{+/+}, PON1^{-/-}, tgHuPON1Q192 and tgHuPON1R192</p> <p>Rationale: The current method of weaning mice at 21 days of age has occasionally resulted in adverse health effects (lethargy, weight loss) in mice of our particular strain (backcrossed B6. 129), because some mice are not fully weaned from the dam by 21 days of age, and have not learned how to use the lixix and/or chew solid food.</p>
4278-02	<p>Weaning Variance: Wean between 21 and 28 days. All transgenic strains to be bred on this protocol have average litter size 6 or less pups per dam. Two types of breeding will be used: trios (two females and one male), and pairs (one female and one male). We will follow University of Washington IACUC Policy on Number of Mice Maintained in a Breeding Cage. Each cage will have no more than 3 adult mice (breeders). If large litters (trio breeding with more than 12 pups) are born, one of the two females with her litter will be separated to a new cage before the pups reach two weeks of age. If a third litter is born prior to weaning of the previous litters, older pups will be separated immediately. When pups of very different ages are present in the cage, the older pups will be either weaned or separated with the dam. If a new litter is born in the cage prior to weaning of the old litter (pups greater than 19 days of age), the old litter will be weaned or separated immediately.</p> <p>Species: Mice, all transgenic strains</p> <p>Rationale: We have been breeding transgenic mouse strains listed on this protocol for several years. In our experience, pups of these strains have reduced viability and benefit from weaning after 21 days of age. Therefore, to increase viability and well-being of pups, we will be weaning pups from all transgenic strains on this protocol between 25 and 28 days of age unless pups need to be separated earlier.</p>
2224-05	<p>Weaning Variance: Late weaning by 5-7 days if pups look smaller than 21 days old or recommended by Vet Services. Late weaning/separating of the first litter by 5-7 days (i.e., at the time the second litter is 5-7</p>

	<p>days old) if a new litter is born in the cage prior to weaning the first litter. Late weaning applies to entire litter. Latest weaning will be 28 days. For standard pair breeding, 1 male, 1 female, and 1 litter will be in the cage. The maximum number of adults and pups that will be in a cage for trio breeding of difficult lines (especially anything on the DBA/2 background or the PiT-2 breeding lines) are 2 females, 1 male, and 12 or less pups.</p> <p>Species: Mice PiT-1 and PiT-2</p> <p>Rationale: Neonatal death is increased within 5-7 days after birth due to a cage disturbance or loud noise/vibration nearby. Early (prior to 5-7 days old) separation of the neonates into a new cage is likely to result in neonate lethality in these lines. A weaning variance is required to reduce stress on the animals and promote breeding with these lines.</p>
2224-07	<p>Weaning Variance: floxed-Runx2, SM22Cre, LDLr-/-, MGP+/- wean 5-7 days late in standard pair breeding.</p> <p>Species: Mouse</p> <p>Rationale: We had better neonate survival if postponed weaning of these lines of mice. Late weaning/separating of the first litter by ~5-7 days (i.e., at the time that the second litter is 5-7 days old) if a new litter is born in cage prior to weaning of the first litter.</p>
2658-01	<p>Weaning Variance: Wean between 28 - 35 days of age. The maximum number of mice in one cage will be 1 adult male, 2 adult females, and 2 litters (12 pups or less in total) less than 35 days old. If a new litter is born prior to weaning of the old litter (19 days or older), the old litter will be weaned or separated immediately.</p> <p>Species: Mice, TCR transgenics, Rag-/-, B cell-deficient, IFN-g-/-, IFN-g receptor-/-, IL-17 receptor-/-, Fas-/-, perforin-/-, GM-CSF-/-</p> <p>Rationale: Many of our TCR transgenic and other immunodeficient strains have pups that are too small to be weaned at 21 days of age. If we leave these pups with their dam until they are 28 to 35 days old, they thrive and do much better.</p>
2013-01	<p>Weaning Variance: Wean at 28 days of age. Breeding cages may house a trio (one male, two females) and no more than 12 pups up to 28 days old. If additional litters are born prior to weaning the older litter(s), the older litter will be moved to a new cage.</p> <p>Species: Mice, various strains: TCR gag RagI-/- X AlbGag RagI-/-; AST X AlbCre; AST GAG Thy1.1; AST X TamCre; SV40-1 Thy1.1 X SV40-1 Thy1.1; SV40-4 Thy1.1+/-; SV40-1 PD1KO; P14 X P14 Thy1.1; P14 Thy1.1+/- Cre Flox+/-; aBeta TCR Thy1.1 Cre Flox; P14Thy1.1+/- clb-b-/-; P14 Thy1.1 X GMIL-2; P14 Thy1.1+/- PD1KO; P14+/- Thy1.1+/- Ly5.1+/- IL10-/- X P14 Thy1.1+/- Cre; P14 Thy1.1 X Ly5.1+/- Cre F/F; P14 Thy1.1+/- TGFBeta+/- Cre; Micro150-/- TCR Thy1.1+/-; TCR Thy1.1+/- clb-b-/- Flox+/- Cre; TCR Thy1.1+/- clb-b-/- PD1KO; aBeta TCR Thy1.1+/- PD1KO; TCR Thy1.1+/- Micro150-/- X Micro150-/-; P14 Thy1.1+/- F/F Cre; SMARTA PD1KO+/-; SMARTA Thy1.1+/- clb-b-/-; POET^2; IL10-/- Cre Ly5.1+/-; OT-1 Ly5.1+/- F/F Cre; OT-1 Ly5.1 Cre IL10-/-; SMARTA Thy1.1; Flox+/- Cre; TRAMP</p> <p>Rationale: Mice of these strains are sometimes relatively small and show increased mortality if weaned at 21 days of age.</p>
3274-02	<p>Weaning Variance: Pups of CaBP1 KO and CaBP2 KO to be weaned between 21 - 28 days of age. For trio breeding, when a new litter is born in the cage prior to weaning of the old litter, the old litter will be separated immediately. For quad breeding, 2 females are separated when apparent that one of them is pregnant.</p> <p>Species: Mice, CaBP1 KO and CaBP2 KO</p> <p>Rationale: Pups with CaBPs targeted genes are still very small at a weaning age of 21 days. In addition these mouse lines produce small litters (~5-6 pups). To increase viability, weaning age will be between 21 - 28 days of age.</p>
4230-01	<p>Weaning Variance: Wean by 28 days, trio with maximum of 12 pups</p> <p>Species: Mice, occasional runts or small litters at 21 days</p> <p>Rationale: Delay weaning for a few days, maximum 1 week, if they are particularly small. No particular strains.</p>
3278-01	<p>Weaning Variance: Wean at 4 weeks of age for LPAB mice and Hem1-/- . Both strains are smaller (<20% smaller) than their littermates at birth and throughout life. We are unsure exactly the mechanisms of dwarfism, but would like permission to wean at 4 weeks of age due to increased weaning stress and mortality after weaning. We will not have more than 2 litters per cage if trio breeding, and will not have more than 12 pups per cage. If we find more than 12 pups, we will remove one litter with their mother until</p>

	<p>the pups are weaned. And, we will separate if an additional litter is born before the old litter is weaned (pups greater than 19 days of age) Species: Mice, LPAB (Fnip1^{-/-}) and Hem1^{-/-} Rationale: Both strains are smaller (<20% smaller) than their littermates at birth and throughout life. We are unsure exactly the mechanisms of dwarfism, but would like permission to wean at 4 weeks of age due to increased weaning stress and mortality after weaning.</p>
4359-01	<p>Weaning Variance: Wean at 28 days of age. Breeding trio (one male, two females) and 1-2 litters of pups up to 28 days old. Female and older litter will be separated until weaning as soon as another litter is born. Species: Mice, Ndufs4, S6k1 knockouts Rationale: Both ndufs4 and s6k1 KO mice can have very small size (<5 grams) at 21 days and may not be able to survive without parental care. Pups will be weaned at 28 days maximum.</p>
4261-01	<p>Weaning Variance: Wean at up to 28 days of age. Number of mice maintained in breeding cages will be per relevant IACUC policy. Species: Mice, PKC KO Rationale: Mice are smaller than normal at 21 days of age of age and therefore need to delay weaning by up to 1 week.</p>
4290-01	<p>Weaning Variance: Wean at 21-28 days of age. For a single breeding pair (1 male plus 1 female per cage) pups to remain with their mothers up to 28 days rather than the standard 21 day. If a new litter is born, the older litter will be weaned immediately. Species: Mice, LmnaKO Rationale: LmnaKO mice have much smaller size and body weight and delayed development, and would benefit from staying with their mothers for slightly longer.</p> <p>Weaning Variance: KDM4Dtet, C57 and FVB mice for neonatal MI project to wean at 3-4 weeks. After surgeries are complete litters will be re-introduced to their foster mother, leaving 1 litter in the cage. If a new litter is born in the cage prior to weaning of the old litter (pups older than 19 days of age), the old litter will be weaned or separated immediately. Species: Mice, KDM4Dtet, C57 and FVB Rationale: Neonates subjected to MI have smaller size and body weight and have delayed development and will benefit from staying with their mothers longer.</p>
2436-12	<p>Weaning Variance: Wean between 24 - 35 days of age. The maximum number of mice in one cage will be one male, 2 females, and 2 litters less than 35 days old; if unusually large litters are born making it so that there are more than 12 pups, 2 females and 1 male, present in the cage at one time, one of the two females with her litter will be separated to a new cage before the pups reach two weeks of age. In the event that a new litter is born to a cage in which pups greater than 19 days of age are present, the older litter will be separated with their dam immediately. If both dams have new litters while older pups are present or we are unable to determine which dam to separate with the older litter, the older pups will be weaned immediately. Species: Mice, Smad3^{-/-}, Rag2^{-/-} double knockout, B6.Smad3^{-/-}, TLR4^{-/-} Rationale: These mice are much smaller than heterozygote or wild type mice and if left with their dam until they are 24 - 35 days old they thrive and do much better.</p>
2022-01	<p>Weaning Variance: Wean between 28 - 35 days of age. Species: Mice, RII mutant, Cbeta, RIIBeta, Calpha, AKAP7 and crosses of these strains. Rationale: Pups are not able to feed themselves at 21 days of age due to genetic mutation. Breeding is done in trios so the cage can have an adult male, two adult females, and two litters less than 35 days of age. If a female has a second litter, one of the females and litter(s) should be removed to another cage.</p> <p>Weaning Variance: After weaning, up to 6 male siblings may be housed together for up to 35 - 42 days of age. Species: Mice Rationale: Animals are genotyped at 21 days of age, and the genotyping process can take from 1 - 2 weeks. Animals that do not have the desired genotyped are euthanized. Housing the males together alleviates the problem that once separated, males cannot be caged together again without fighting.</p> <p>Tumor growth in RIalpha KO and mutant mice usually happens after 1 year and mice are euthanized prior to that age.</p>
2856-01	<p>Weaning Variance: Wean at 30 days of age. Breeding cages can contain: 1 adult male, 2 adult females and</p>

	<p>2 litters up to 30 days old. However, see rationale below.</p> <p>Species: Mice, any strains that may have retarded growth but need to be kept into adulthood, especially Bax^{-/-}, Bif-1^{-/-}, Bif-1c (added in version 74).</p> <p>Rationale: As newborn animals in our mouse colonies, especially those of knockout strains, tend to show somewhat retarded growth we request a weaning variance in which weaning is delayed, when necessary, until 30 days after birth in order to reduce post-weaning deaths due to premature weaning. Mating cages of these knockout mouse strains will have the regular format of 1 male-2 females-2 litters (or the equivalents of 5 adults). However, these mice are not as fertile as their wild-type counterparts and their litter sizes tend to be smaller. Also, they are mated so that 2 litters are born at least one week apart. Thus, there is little chance of having 2 litters of similar age and ordinary litter size (8-10 pups) present in a cage at any one time. In such rare occasions, we will make an effort to reduce the number of pups by euthanizing unnecessary pups (typically male mice in our case) halfway toward weaning. As soon as the pups reach the body size for safe weaning, they will be weaned. This weaning variance applies to any mice that are born small but need to be kept into adulthood, but the following strains tend to require the variance more frequently: p53^{-/-}, Bax^{-/-}, Bif-1^{-/-}, and Bif-1c (added in version 74).</p>
4246-01	<p>Weaning Variance: Wean at 28 days of age.</p> <p>Species: Mice, double knockout mice</p> <p>Rationale: Weaning at a later age is beneficial for mice with reduced viability. Double knockout mice often have reduced viability. Gel and diet on the bottom of the cage will be provided at weaning and the sign "Gel/Toggle" attached to the front of weaned cage. To avoid tramping newborn pups by previous litter, cages with P15-P18 pups will be checked for pregnant female(s) and pregnancy stage. Pups will be weaned earlier (starting at 19 days) if next litter is to be born within 1-2 days.</p>
2183-02	<p>Weaning Variance: Wean at 28 days of age.</p> <p>Species: Mice, Th fs/fs</p> <p>Rationale: These mice are small and need to be left with their mothers a little longer than normal (but no longer than 28 days).</p>
2650-07	<p>Weaning Variance: Wean at 28 days of age for apo E^{-/-} strain.</p> <p>Species: Mice, apo E^{-/-}</p> <p>Rationale: The pups of the apo E^{-/-} mice are smaller than normal C57Bl/6.</p>
2650-08	<p>Weaning Variance: Wean at 28 days. Mice are bred as 2 females and 1 male. If a dam drops a second litter, wean the older pups immediately.</p> <p>Species: Mice apoE^{-/-}</p> <p>Rationale: Mice are undersized at 21 days and do not thrive well if weaned.</p>
2456-06	<p>Weaning Variance: Wean at up to 27 days of age.</p> <p>Species: Mice, PPARgNKO and WT</p> <p>Rationale: Weaning time is an experimental variable (20 vs 27 days). PPARg controls body weight in a manner that is dependent on time of weaning might suggest that this receptor is an important factor in the post weaning development of neurocircuits involved in the regulation of body weight, food intake and energy homeostasis. Assess post-weaning body and food intake in PPARgNKO and WT mice both after a normal weaning time (20 days) and after prolonged weaning (27 days).</p>
4196-01	<p>Weaning Variance: Wean at 28 days of age. Cage will contain 2 adult females and 1 adult male and 2 litters of up to 28 days of age.</p> <p>Species: Mice, AKAP150 and others.</p> <p>Rationale: Some strains of mice (e.g., AKAP150, AKAP220, Wave1KO, crosses of these) are undersized for their age.</p>
2011-21	<p>Weaning Variance: Wean AC3^{-/-} mice at 28 days of age. Breeding trio (one male, two females) and 1-2 litters of pups up to 28 days old. We request leaving these pups with their dam until they are 28 days old, we will wean these mice at 28 days with 2 normal littermates and 1-2 runts. We will reduce the litter size to 3-4 pups per litter at 21 days. . In the event that a new litter is born to a cage in which pups greater than 21 days of age are present, the older litter will be separated with their dam immediately. If a dam has a new litter prior to weaning its older pups then the older pups will be weaned immediately.</p> <p>Species: Mice (strain AC3^{-/-})</p> <p>Rationale: The AC3^{-/-} mice are runts and too small to be weaned at 21 days of age. We usually get 1-2 runts per litter and will keep the runt pups with their two normal littermates until 28 days; the normal littermates are to be used as controls for the runt litter mates.</p>

3410-01	<p>Weaning Variance: Weaning age up to 28 days with no more than 3 adult mice (2 females and one male) and two litters (? 12 pups) in a cage at any time. When there are two litters in a cage at the same time the older litter will not be > 18 days old. When a litter is born in a cage that already has a litter > 18 days either the older litter will be weaned immediately (if 21 days or older and appear to be doing well) or the younger litter will be removed and sacrificed immediately or before the older litter reaches 19 days old.</p> <p>Species: Mice on background strain of C57BL/6;129Sv</p> <p>Rationale: All of the protocol's mice are on a background strain of C57BL/6;129Sv. This inbred strain is known to have small litters of slow developing pups with relatively long inter-litter intervals.</p>
2199-09	<p>Weaning Variance: Wean at 28 days of age with 1 adult male and 2 adult females in breeding cage.</p> <p>Species: Mice, Deafwaddler and quivering strains, Potassium channel mutants Kcna1KO, Kcan1EA1, Kv1.2KO PMCA2 mutants Atp2b2dfw, Atp2b2dfw2J, Atp2b2dfwi5, Atp2b2Tg-dox, Atp2b2V586M β-spectrin4 mutants Spnb4qv, Spnb4qv4J Claudin9 mutant Cldn9Tg-dox</p> <p>Rationale: Several of our strains have smaller pups and reduced litter sizes often of only 4-5 pups. These pups do not develop motor skills sufficient for them to be weaned at 21 days. We find that maintaining stable trios (1 adult male and 2 adult females) and allowing the pups to stay with their moms until 28 days for weaning helps the pups be able to reach/eat the food and water. Given the small pups and reduced litter size this variance should not cause overcrowding.</p>
3041-04	<p>Weaning Variance: Wean at 28 days of age with 1 adult male and 2 adult females in breeding cage.</p> <p>Species: Mice, Deafwaddler and quivering strains, Potassium channel mutants Kcna1KO, Kcan1EA1, Kv1.2KO PMCA2 mutants Atp2b2dfw, Atp2b2dfw2J, Atp2b2dfwi5, Atp2b2Tg-dox, Atp2b2V586M β-spectrin4 mutants Spnb4qv, Spnb4qv4J Claudin9 mutant Cldn9Tg-dox</p> <p>Rationale: Several of our strains have smaller pups and reduced litter sizes often of only 4-5 pups. These pups do not develop motor skills sufficient for them to be weaned at 21 days. We find that maintaining stable trios (1 adult male and 2 adult females) and allowing the pups to stay with their moms until 28 days for weaning helps the pups be able to reach/eat the food and water. Given the small pups and reduced litter size this variance should not cause overcrowding.</p>
2488-10	<p>Weaning Variance: Wean at 28 days of age.</p> <p>Species: Mice, Rag 1^{-/-}, mdx and alpha7 integrin</p> <p>Rationale: Because of the small size of these animals, there is an increase in deaths post-weaning if they are weaned at 21 days of age. The animals at 28 days of would be a more typical size for weaning.</p>
4249-01	<p>Weaning Variance: Wean Drd1a-null mice at up to 28 days of age. Breeding trio (one male, two females) and 1-2 litters of pups up to 28 days old may be in the cage at any one time. If additional litters are born prior to weaning the older litter(s), the new litter will be removed.</p> <p>Species: Mice, Drd1a-null strain</p> <p>Rationale: These mice are smaller than littermate controls and require one extra week before weaning.</p>

Protocol Number	Temperature Variance
2070-06	<p>Temperature Variance: Mice caged from 1-6 hours at 4 degrees Celsius</p> <p>Species: Mice</p> <p>Rationale: Used to provide a physiological delivery of adrenergic stimulation for study of body temperature control.</p>
3104-01	<p>Temperature Variance: 5 degrees C for 4 hours, 14 degrees C for 3 days</p> <p>Species: Mice</p> <p>Rationale: For acute cold stress measurements, mice will be placed inside the test chamber and ambient temperature will be reduced from 22 degrees C to 5 degrees C over a 60 minute period. The temperature will be maintained at 5 degrees C + or - 0.5 degrees C for 240 minutes and then raised back up to 22 degrees C over the next 60 minutes. Each cohort of mice will be subjected to a single acute cold stress over a 3-day period. Following recovery (at least 5 days), an additional second chronic cold stress will be induced by reducing ambient temperature to 14 degrees C over a 60-minute period. The temperature will be maintained at 14 degrees C + or - 0.5 degrees C for 3 days and then raised back to 22 degrees C over the next 60 minutes. These measurements are necessary to determine the effect of CLA on thermogenesis in a mouse model of diet-induced obesity.</p>
4377-01	<p>Temperature Variance: Elevated housing temperatures</p> <p>Species: Zebrafish</p>

	Rationale: Exposure of larval and adult fish to temperatures ranging from 29 - 33 degrees C to inactivate a particular immune receptor.
3328-05	Temperature Variance: Use of a cooling cap for 72 hours Species: <i>Macaca nemestrina</i> Rationale: The group is studying hypothermia as part of a treatment for brain injury during or immediately following birth.
4118-01	Temperature Variance: 5 degrees C for 4 hours. Species: Mice Rationale: We will conduct the varied temperature experiment using environmental test chambers (incubators). All the energy homeostasis measurements can be performed within the two incubators by placing the Sable systems metabolic cages. For experiments examining the metabolic response to cold adaptation, the environmental chambers will then be set at 5°C and allowed to reach that temperature overnight. The following day, mice will be placed in individual metabolic cages that contain bedding that are housed within the environmental chamber at 5°C. Following four hours of exposure at 5°C, animals will be removed and immediately sacrificed.
2057-01	Temperature Variance: Species: Zebrafish Rationale: For induction of transgene expression zebrafish will be heat-shocked for 1 hour per day for the length of the experiment (up to 42 days post transection) by raising the temperature of their water from 28°C to 37°C slowly over 15-30 minutes, then returning the fish to 28°C water. The lab has found that heat-shocking transgenic fish one time per day for the length of the experiment for a duration of 1 hour leads to adequate induction of the transgene as measured by GFP expression. Heat-shock will not be administered prior to completion of the 5-day post-operative monitoring procedure.
4246-01	Temperature Variance: Housing at 29 +/- C Species: Mice Rationale: To provide thermoneutral condition because the behavior and physiology studied are optimal at this temperature.
2456-06	Temperature Variance: Thermoneutral Species: Mice, Rats Rationale: Animals undergo a series of metabolic experiments. Normally, room temperature is kept between 68° and 78° F. However, to eliminate the possibility that a defect in the maintenance of body temperature is contributing to the enhanced energy expenditure in rodents of a particular genotype, the same metabolic experiments need to be performed under thermoneutral conditions. This experiment is performed routinely in metabolic chamber systems and does not harm the animal. Room temperature will be manually raised from 76° to 90° F with a 4 ~5 degree rise per every 48 hour. Animals will be placed in the metabolic chambers for a total of 192 hours. OR Temperature Variance: Heat exposure Species: Mice, Rats Rationale: Metabolic experiments. Ambient temperature (Ta) of the chamber will be increased from 22°C to as high as 38°C over a period of 60 min. Ta will be maintained at 38 ± 0.5°C for 120 min and then returned to 22°C over the next 60 min. Each cohort will be tested in the incubators over a 3-day period. Animals will be monitored every 15 minutes throughout the duration of the heat exposure. OR Temperature Variance: Cold stress Species: Mice, Rats Rationale: Metabolic experiments. Ambient temperature (Ta) will be reduced from 22°C to 7°C over a 60-min period. A Ta value of 7 ± 0.5°C will be maintained for 120 min and then will be raised back to 22°C over the next 60 min. Each cohort will be subjected to cold stress over a 3-day period. Animals will be monitored every 15 minutes throughout the duration of the heat exposure. OR Temperature Variance: Cool Temperature Species: Mice (C57Bl/6 and ob/ob)

	<p>Rationale: Metabolic experiments. Ambient temperature (Ta) will be reduced from 22°C to 14°C. A Ta value of 14°C will be maintained for up to 1 week and then will be raised back to 22°C. This cool exposure time is less than that previously reported in the literature (2 weeks).</p>
2456-06	<p>Temperature Variance: Thermoneutral Species: Mice, Rats Rationale: Animals undergo a series of metabolic experiments. Normally, room temperature is kept between 68° and 78° F. However, to eliminate the possibility that a defect in the maintenance of body temperature is contributing to the enhanced energy expenditure in rodents of a particular genotype, the same metabolic experiments need to be performed under thermoneutral conditions. This experiment is performed routinely in metabolic chamber systems and does not harm the animal. Room temperature will be manually raised from 76° to 90° F with a 4 ~5 degree rise per every 48 hour. Animals will be placed in the metabolic chambers for a total of 192 hours.</p> <p>OR Temperature Variance: Heat exposure Species: Mice, Rats Rationale: Metabolic experiments. Ambient temperature (Ta) of the chamber will be increased from 22°C to as high as 38°C over a period of 60 min. Ta will be maintained at 38 ± 0.5°C for 120 min and then returned to 22°C over the next 60 min. Each cohort will be tested in the incubators over a 3-day period. Animals will be monitored every 15 minutes throughout the duration of the heat exposure.</p> <p>OR Temperature Variance: Cold stress Species: Mice, Rats Rationale: Metabolic experiments. Ambient temperature (Ta) will be reduced from 22°C to 7°C over a 60-min period. A Ta value of 7 ± 0.5°C will be maintained for 120 min and then will be raised back to 22°C over the next 60 min. Each cohort will be subjected to cold stress over a 3-day period. Animals will be monitored every 15 minutes throughout the duration of the heat exposure.</p> <p>OR Temperature Variance: Cool Temperature Species: Mice (C57Bl/6 and ob/ob) Rationale: Metabolic experiments. Ambient temperature (Ta) will be reduced from 22°C to 14°C. A Ta value of 14°C will be maintained for up to 1 week and then will be raised back to 22°C. This cool exposure time is less than that previously reported in the literature (2 weeks).</p>
3233-05	<p>Temperature Variance: Thermal seizure induction Species: Mice Rationale: Thermal seizure induction - core temp potentially raised through 37C - 42.5C - maximum duration 22 minutes outside of normal housing temperatures.</p>
3041-04	<p>Temperature Variance: Species: Mice Rationale: Mice in MPTP experiment in K017B with a temperature range of 68-81 degrees F before and after MPTP injection with an average temperature setting of 76 degrees F. The average setting falls within the range of the Guide (68-79) but there is a chance that the daily high may exceed the Guide. Temperature should not be reset unless it exceeds 81 degrees. Sensaphone still alarms at 82 degrees F. The purpose is that MPTP-treated animals have better survival if kept warmer. Controls must be kept at the same temperature as MPTP animals.</p>
Protocol Number	IACUC Policy Variance
2013-01	<p>Policy Variance: Breeding scheme variance Species: Mice Rationale: Due to the difficulty breeding some transgenic strains, some breeder cages will have three females and two sibling males. Pregnant females will be moved to a new cage before giving birth. Variance: House 6 mice < 25 grams per cage for a 4 week period between weaning and genotyping. Species: Mice, see strains under Weaning Variance above</p>

	<p>Rationale: The strains of mice being genotyped are less than 25 grams so technically 6 animals per cage is within the requirements in the GUIDE.</p>
4336-01	<p>Policy Variance: Loss of body weight >30% in bleomycin lung injury model mice where IGF-1 neutralizing antibody is studied.</p> <p>Species: Mice</p> <p>Rationale: IGF anti-body treated mice had significant weight loss (20%-25%), but did not exhibit other signs of distress, such as moderate to severe tachypnea or dyspnea, or hunched posture. In consultation with Veterinary Services we opted to carefully monitor the mice. We found that despite the weight loss, our IGF-1 antibody treated mice had less than anticipated spontaneous death rate and actually found an improvement in measurements of pulmonary fibrosis and improved survival. Significant weight loss associated with prolonged survival is well-described in other animal models in which the IGF pathway is blocked. Therefore, in the bleomycin lung injury model where IGF-1 antibody is studied, weight loss does not correlate with respiratory distress or increased mortality.</p>
2856-01	<p>Policy Variance: Breeding cages with 1 male and 4 females.</p> <p>Species: Mice of strains used for embryonic or neonatal neuronsm esoecuakkt p53 and C57BL/6J.</p> <p>Description: Timed mating to obtain newborn pups for culture work.</p> <p>Rationale: We request to breed 1 male/4 females following situations and reasoning.</p> <p>(1) To do culture work using newborn pups, it is desirable to know when pups are born and available in order to schedule experiments in advance. Also, for establishing embryonic neuron cultures, timed mating is imperative. For these purposes, a male will be put with 4 females in a cage and plugs will be checked on the following days. Animals will be separated to conform to one of the options allowed for a breeding cage before pups are born. Newborn pups will be used for experiments as needed and, when necessary, kept for colony maintenance purposes. Surplus pups will be euthanized. Based on our experience in the past, four females all become pregnant sooner or later in most cases and therefore there seems to be no adverse effect of this breeding format on breeding behavior and efficiency. Thus, the harem mating (1 male/4 females) is an efficient breeding scheme, consistent with effective use of limited resources (the number of animals, space and budget) without sacrificing performance. This specifically applies to the p53 wild-type colony and C57BL/6J. The breeding cages thus started as harem mating may eventually be kept for colony maintenance in one of the formats (typically as trio breeding) allowed for a breeding cage.</p> <p>(2) Regular breeding to maintain certain strains. Some other strains we maintain are not necessarily as prolific as we would like. Some females are less efficient and often nearly infertile. For these strains, we would like to initially set up 1 male/4 females breeding cages. Once pregnancies are identified, visibly non-pregnant mice will be removed from the cage before pups are born so that the number of remaining animals conforms to one of the options allowed (trio or quad breeding). This way, we can select females that are readily impregnated and therefore can be used as reliable females to maintain the lines for months to follow. The past experience showed that those females that were not impregnated in the first two weeks or so of mating either were inefficient or did not get impregnated at all in the following weeks of continuing breeding. Thus, the initial inefficiency seen with some females does not appear to be caused by the breeding condition of 1 male/4 females format, but rather is due to low fertility inherent to those specific females. This applies to Bax KO line, Bif-1 KO lines, Alzheimer.s disease model strain and mito-CFP transgenic line.</p> <p>(3) For difficult-to-breed strains. This is specific to our p53-KO line. Females of this strain show very limited fertility (~50% at best). Fertile females are relatively reliable breeders, but others are nearly completely infertile. Housing those infertile females in the trio format is inefficient in every way. Harem mating (1 male/4 females) allows efficient selection of fertile females without sacrificing the performance of those fertile females. In a typical setting, once pregnant females approach due dates, non-pregnant females will be removed and, depending on their mating history, either euthanized or put in another harem breeding cage. Ongoing breeder cages with fertile females will be in one of the options allowed (standard/trio/quad).</p> <p>Note: The one male/four female format we are requesting here is not for ongoing breeding. Regular breeder cages will strictly conform to one of the options currently allowed.</p> <p>Trio breeding will follow the IACUC guideline Number of Mice Maintained in Breeding Cages as follows with some modifications [in brackets].</p> <p>(a) Trio breeding can be used only for inbred strains or genetically modified mice where small litters (average 6 pups or less per litter) or poor breeding efficiency is seen. [Trio breeding will also be used for stains/lines that are expected to have litters of more than 6 pups but by following the next rule with our modification (below in brackets)].</p> <p>(b) If unusually large litters (more than 12 pups, 2 females and 1 male) are born, one of the two females with</p>

	<p>her litter must be separated to a new cage before the pups reach two weeks of age. [If large litters are born in fertile strains/lines, animals will either be separated according to the rule or extra pups will be euthanized to keep the number of pups no more than 12. This is a reasonable approach for us because we need to keep more females than males for mating purposes and all pups do not have to be weaned and kept for maintaining the strains/lines. Thus, excess pups, especially males, can be euthanized to meet the no more than 12 pups rule. Also, for the strains/lines from which we obtain newborn pups for culture work, pups will be used up on the day of birth and overcrowded conditions will not ensue.]</p> <p>(c) If a new litter is born in the cage prior to weaning of the old litter (pups greater than 19 days of age), the old litter will be weaned or separated immediately. [When older pups are small and our weaning variance is applicable, the weaning/separation of older pups will occur when they reach the size of normal 19 day-old pups.]</p> <p>(d) When pups of very different ages (i.e., 19-20 days old versus newborn) are present in the cage, the older pups must be either weaned or separated with the dam. [When older pups are small and our weaning variance is applicable, the weaning/separation of older pups will occur when they reach the size of normal 19 day-old pups.]</p>
2950-01	<p>Policy Variance: Food on floor of cage Species: Mice and rats Rationale: Surgical implants interfere with hopper and smaller animals thrive better due to individual differences in growth rate</p>
2048-02	<p>Policy Variance: Permissible Weight Loss: Species: Mice and Rats Rationale: We are approved to administer aminoglycoside antibiotics to rats and mice. AG administration often results in a temporary drop in weight. Animals typically recover their weight a week or two following treatment. During treatment, animals are weighed and monitored daily. If weight loss is observed, animals are given nutritive supplements and saline injections to assist their recovery. We wish to adjust our current euthanasia criteria to allow for veterinary consultation when an animal's weight loss approaches 20%. In this way, if an animal is showing signs of improvement and meets with veterinary approval, supportive measures may be continued and the animal may remain in the study. If rats reach 20% weight loss, Veterinary Services will be contacted to determine if additional supportive care is appropriate or euthanasia is warranted on a case-by-case basis. Between 20-30% weight loss, supportive care and monitoring will be done in conjunction with Vet Services, with ongoing assessment to determine if the rat is at a humane endpoint. Rats will be euthanized if they reach 30% weight loss despite intensive supportive care.</p>

Protocol Number	Cage Variance
2070-06	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
3104-01	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
3104-05	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
3361-01	<p>Cage Variance: Species: MICE Description: CLAMS metabolic cages are smaller than Guide recommendations. Rationale: Animals are housed in these cages up to 72 h for indirect open-circuit calorimetry.</p>
3298-02	<p>Cage Variance:</p>

	<p>Species: Mice Description: Mice are housed for 24-48 hours in CLAMS cages for metabolic screening. The CLAMS cages are 4" in height. Rationale: The CLAMS caging system is necessary to perform a comprehensive characterization of energy homeostasis.</p>
3437-01	<p>Cage Variance: Species: Mice Description: Mice are housed in the Comprehensive Lab Animal Monitoring System (CLAMS) to measure total energy expenditure, food and water intake, and physical activity for up to 72 hours. Rationale: The CLAMS indirect calorimeter is preferred to that provided by Sable Systems (which has adequate cage dimensions) depending on the study design and the measures required to perform a comprehensive characterization of the energy homeostasis phenotype of the animal. The CLAMS cage does not contain bedding, which allows for collection and quantification of urine and feces over designated time periods. The feeding apparatus is equipped to continuously measure food intake allowing the calculation of the size of the meal, the number of meals consumed, and the time between meals, which are important measures in the study of obesity and insulin resistance. The Sable System has a flawed hopper design, which results in excess food spillage into the cage; therefore, the optimal collection of metabolic data requires use of the CLAMS unit.</p>
4360-01	<p>Cage Variance: Requesting a housing variance so that we may use this specialized tether compatible caging that measures 45in2. Species: Guinea pig Description: Housing guinea pigs in a smaller cage for 18-42 hr Rationale: This cage size is compatible with the tether system being used.</p>
4378-01	<p>Cage Variance: Species: Mice Description: Metabolic screening, as described in this protocol, may involve the use of either the Sable or CLAMS systems. The CLAMS cages for mice do not meet the federal regulation, as stated in the Guide, for cage height to be at least 5". Cage height is 4" for the CLAMS mouse cages. We request a cage-size variance for the use of the CLAMS mouse cages. Rationale: The CLAMS indirect calorimeter is preferred to that provided by Sable Systems (which has adequate cage dimensions) depending on the study design and the measures required to perform a comprehensive characterization of the energy homeostasis phenotype of the animal. The CLAMS cage does not contain bedding, which allows for collection and quantification of both urine and feces over designated time periods. The feeding apparatus is equipped to continuously measure food intake allowing the calculation of the size of the meal, the number of meals consumed, and the time between meals. This information can provide scientific insight into the biological mechanism(s) that cause obesity. Unfortunately, with the Sable Systems feeder, the hopper has design flaws resulting in excess food spillage into the cage, making this analysis impossible. In addition, the CLAMS unit is configured best to make measurements of core body temperature using an implanted telemetry transponder.</p>
4118-01	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
2174-23	<p>Cage Variance: Species: Mice Description: Metabolic screening, as described in this protocol, may involve the use of either the Sable, or CLAMS systems. The CLAMS cages for mice do not meet the federal regulation, as stated in the Guide, for cage height to be at least 5". Cage height is 4" for the CLAMS mouse cages. Rationale: The CLAMS indirect calorimeter is preferred to that provided by Sable Systems depending on the study design and the measures required to perform a comprehensive characterization of the energy homeostasis phenotype of the animal. The CLAMS cage does not contain bedding, which allows for collection and quantification of both urine and feces over designated time periods. The feeding apparatus is equipped to continuously measure food intake allowing the calculation of the size of the meal, the number of meals consumed, and the time between meals. This information can provide scientific insight into the biological mechanism(s) that cause obesity. Unfortunately, with the Sable Systems feeder, the hopper has</p>

Appendix D

	design flaws resulting in excess food spillage into the cage, making this analysis impossible. In addition, the CLAMS unit is configured best to make measurements of core body temperature using an implanted telemetry transponder.
4130-01	<p>Cage Variance: Species: Mice Description: Mice are housed for a period of 48 hours in CLAMS cages for calorimetry measurements. The CLAMS cages are 4” in height. Rationale: The CLAMS caging system is the preferred calorimetry system to best measure food consumption. Accurate whole-body measures of energy expenditure and food intake are critical to understand the bioenergetics underlying increased exercise performance.</p>
2022-01	<p>Cage Variance: Species: Mice Description: CLAMS indirect calorimetry cages do not meet minimal cage height and a variance is requested for these units. The CLAMS cage height is 4 in. and the height requirement in the Guide is 5 in. Rationale: Individual measurement of calorimetry and intakes must be done in these cages, as they provide more accurate measurements than other cages that do meet Guide standards for height.</p>
2183-02	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
3062-01	<p>Cage Variance: Species: Chickens Description: For chicks <3 weeks old, using recommendations of Guide for Care and Use of Agricultural Animals in Research and Testing instead of Guide for Care and Use of Laboratory Animals, caging will be: males - floor space at 20" squared each; females - floor space at 15" squared each. Rationale: This will allow the use of only one brooder. This improves chick care because it provides an open brooder to rotate chicks between brooders while they are cleaned.</p>
2048-02	<p>Cage Variance: Species: Chicks <3 weeks old Description: Male chicks - floor space at 20 square inches each; Female chicks - floor space at 15 square inches each. Using recommendations of Guide for Care and Use of Agricultural Animals in Research and Testing (3rd Edition), instead of Guide for Care and Use of Laboratory Animals. Rationale: This will allow the use of only one brooder. This improves chick care because it provides an open brooder to rotate chicks between brooders while they are cleaned.</p>
4369-01	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>
2456-06	<p>Cage Variance: Species: Mice Description: CLAMS caging for mice has a height of 4" Rationale: Metabolic screening is done in the CLAMS caging system because of the superior homeostasis information that it provides.</p>



UNIVERSITY OF WASHINGTON
INSTITUTIONAL ANIMAL CARE AND USE COMMITTEE

January 21, 2016

To: Institutional Animal Care and Use Committee Members

From: Tena Petersen
University of Washington
Manager, Regulatory Programs
Institutional Animal Care and Use Committee
Box 357160

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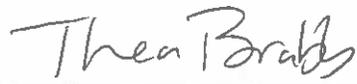
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Manager of Program Operations
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tena@uw.edu

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Signature



Printed Name



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Signature

JEANOT MUSTER

Printed Name



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 01-27-16

Signature

Michelle Newman
Printed Name



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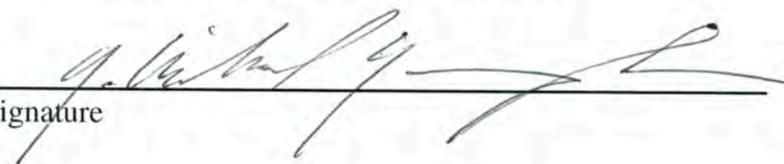
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Signature

G. Michael Gough
Printed Name



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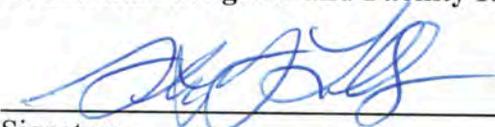
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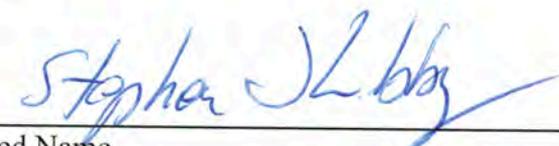
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Jane Sullivan

Signature

JANE SULLIVAN

Printed Name



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Margaret R. Byers

Signature

MARGARET R. BYERS

Printed Name



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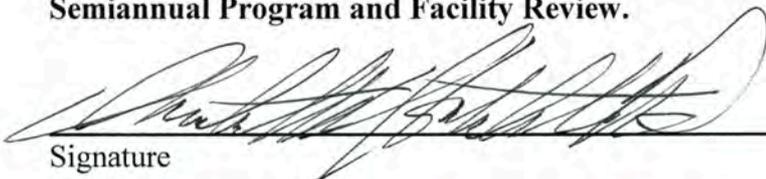
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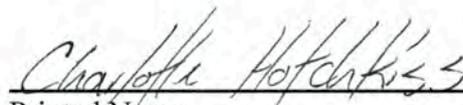
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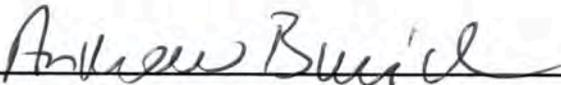
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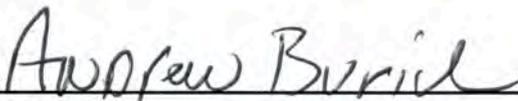
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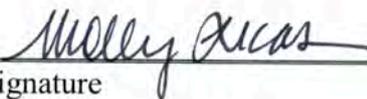
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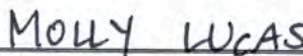
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Aubrey Schoenleben

Printed Name