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Establishment of Blood Analyte Intervals for Laboratory Mice and Rats by Use of a Portable Clinical Analyzer

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Portable clinical analyzers are currently used in human and veterinary medicine for diagnostic testing and blood monitoring; however, normal values for mice and rats of varying genetic backgrounds have not previously been reported. Blood was collected from unanesthetized mice (n = 131) and rats (n = 76) into lithium heparin tubes for analysis using E6+ cartridges for the portable analyzer. Results of glucose, blood urea nitrogen (BUN), sodium, potassium, chloride, hematocrit, and hemoglobin were compared to published ranges provided by a contract diagnostic laboratory. Analyzer ranges were computed as the mean ± 2 standard deviations of the test samples, such that approximately 95% of tested animals would fall within the resultant range. The degree of overlap between analyzer and published ranges, or the percentage of the published range contained within the analyzer range, was calculated for all analytes. For mice, the ranges of 5 of 7 analytes had more than 57% overlap; for rats, ranges for 6 of 7 analytes had over 65% overlap. After the establishment of normal ranges, the analyzer was used to confirm hyperglycemia in Type I diabetic mice and elevated BUN in rats with induced glomerulosclerosis. The portable analyzer can be a valuable screening tool for both phenotyping and clinical care of rodents, with potential for investigations of both spontaneous and experimental disease in laboratory rodents.

Abbreviations: BUN, blood urea nitrogen; NIH, National Institutes of Health; SD, Sprague-Dawley

The field of laboratory animal medicine needs rapid, high-throughput, and economical methods to clinically evaluate and phenotype individual rodents. Particularly for rodent clinical pathology assessments, appropriate medical equipment must be available to process a cadre of tests using minimal blood sample volumes. Portable clinical analyzers have been used extensively in human medicine[1,6,11] and in traditional clinical veterinary and exotic animal practice,[9,14,15,21,22] especially for point-of-care and emergency and critical care cases. The benefits of this instrument include automatic calibration and quality control, low to no maintenance, convenience, and versatility. Small sample volumes of approximately 3 drops (typically whole blood collected in lithium heparin anticoagulant) can yield multiple test results in 2 min. A wide range of blood parameters, encompassing routine electrolytes, glucose, clotting times, acid-base status, and blood gases, can be analyzed depending upon the portfolio of the selected cartridge. In addition, the analyzer has been documented as reliable, robust, and simple for nonlaboratory personnel to operate.[17,22]

In veterinary medicine, reference intervals for portable clinical analyzers have been reported for dogs, cats, and horses;[10] however, similar reference intervals are not available for mice and rats. Within the specialty veterinary field of laboratory animal medicine, a portable analyzer was recently used to validate pathophysiologic markers of disease in a mouse model of disseminated candidiasis.[19] In addition, the same handheld analyzer has been compared with large-scale industrial analyzers for mouse samples.[20] For rats, the same analyzer has been used to measure the influence of ethanol administration on blood levels of ionized calcium and pH.[12,13] Measurement of pH and partial and total CO_2 in rats has been compared between the portable analyzer and a large-scale radiometer.[8]

For our purposes, we viewed a portable clinical analyzer as an optimal tool for antemortem clinical and phenotypic assessments in individual laboratory rodents, due to the small volume of sample required for tests and the ability to transport the analyzer to decentralized areas for 'cage-side' clinical evaluations. Our objective in this project was to establish normal ranges for a panel of 7 chemistry, hematologic, and electrolyte markers in clinically normal laboratory mice and rats using a portable analyzer. We also sought to compare the portable analyzer's values with published range intervals available for these species. After establishing normal ranges of values for laboratory rodents, we then used the analyzer to confirm alterations in specific disease markers of experimental rodent models.

Methods and Materials

Animals. All tested animals were cared for in compliance with the *Guide for the Care and Use of Laboratory Animals*[18] under protocols approved by the University Committee on the Use and Care of Animals. Facilities housing the animals were accredited by the Association for Assessment and Accreditation of Laboratory Animal Care, International.

Mice (*Mus musculus*). BALB/cAnNCrlBR mice (n = 44; 29 female, 15 male), C3H/HeNCrlBR mice (n = 49; 35 female, 14 M), and 129/SvPasIcoCrlBR mice (n = 38; 21 female, 17 male) were evaluated to establish the reference ranges. Mice ranged in age from 17 to 345 d and were housed on ventilated racks in microisolation caging containing corncob bedding (Bed-o'cobs, The Andersons, Maumee, OH). Cages were provided with automatic water and ad libitum chow (LabDiet 5001, PMI International, Brentwood, MO). The blood samples were

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harvested from individual animals prior to euthanasia by CO_2 inhalation.

Male mice (n = 4; age, 135 to 184 d) on a mixed genetic background of 129.SVJ, C57BL/6J, and FVB (STOCK-Tg[H2-Cre]Glut4$^{lox/lox}$) enrolled in a diabetes study were evaluated for blood glucose levels, in both fasted (for 4 h) and nonfasted states. These mice are models of type I diabetes mellitus and are used to study the role of glucose transporters in diabetic kidney disease.[3] After induction of disease with streptozotocin (40 mg/kg intraperitoneally daily for 5 consecutive days), the laboratory staff routinely verifies hyperglycemia (defined by values exceeding 250 mg/dl) in mice by use of a human glucose meter (Accu-Chek Advantage, Roche Diagnostics, Basel, Switzerland). Diabetic mice were housed in static microisolation caging with water bottles; all other aspects of husbandry are similar to those conditions described for clinically normal mice. Animals were bled twice as part of their serial evaluation for hyperglycemia and remained in the colony after sampling.

At our institution, mice undergo routine health testing. Mice were free of pinworms (*Syphacia* and *Aspiculuris* spp.) by cecal exam and were antibody-negative for mouse hepatitis virus, mouse parvoviruses, rotavirus, *Ectromelia* virus, Sendai virus, pneumonia virus of mice, Theiler murine encephalomyelitis virus, reovirus, *Mycoplasma pulmonis*, lymphocytic choriomeningitis virus, mouse adenovirus, and polyomavirus.

***Rats* (Rattus norvegicus).** Sprague-Dawley (SD) rats (n = 50; 28 female, 22 male) and National Institutes of Health (N:NIH) rats (n = 26; 6 female, 20 male) were evaluated to establish the reference ranges. Rats ranged in age from 28 to 543 d and were housed on ventilated racks in microisolation caging containing corncob bedding (The Andersons) and provided ad libitum water and chow (LabDiet 5001, PMI International). Rats were returned to their cages after sample collection.

F344-Tg(DTR)C354Wig rats (n = 10; 7 female, 3 male) transgenic for the human diphtheria-toxin receptor, which is driven by a podocyte-specific promoter within the kidney, were evaluated. To produce the model, rats are inoculated with varying doses of diphtheria toxin, to regulate podocyte depletion and induce glomerulosclerosis in adult rats with normally developed kidneys.[25] Control animals are inoculated intraperitoneally with saline (10 ml/100 g body weight; 0.9% for injection). The laboratory routinely confirms induced disease by measurement of elevated urine protein:creatinine (defined by values greater than 1.5) in these animals. These transgenic rats (experimental, 9; control, 1) were housed under husbandry conditions similar to those described for clinically normal rats.

Rats also undergo routine health assessments. They were free of pinworms by cecal exam and were antibody-negative for sialodacryoadenitis virus, rat parvoviruses, Sendai virus, pneumonia virus of mice, Theiler murine encephalomyelitis virus, reovirus, *M. pulmonis*, lymphocytic choriomeningitis virus, and mouse adenovirus.

Equipment. Equipment used for blood collection and processing of the samples included a lithium heparin 1.3-ml tube (D-51588, Sarstedt Aktlengesellschaft and Company, Nümbrecht, Germany), a disposable plastic pipette, the disposable single-use cartridge (E6+ Mini Urgent Care cartridge), the portable analyzer (i-STAT Portable Clinical Analyzer, Abbott Point-of-Care, East Windsor, NJ), and the infrared printer (Hewlett Packard, Palo Alto, CA; Figure 1).

Blood collection. Blood samples were collected from clinically normal animals that were enrolled in various noninvasive protocols (for example, surplus animals or those used for training) or were experimentally naïve.

The submandibular vein was used for collection of blood samples from unanesthetized mice. Mice were manually restrained by a 1-handed grip, and a disposable lancet (MEDIpoint International, Mineola, NY) was used to prick the submandibular vein. Unanesthetized rats were restrained in disposable restraint bags that allowed access to the saphenous vessel. A hindleg was isolated, and a scalpel blade used to remove hair over the vessel. A small amount of ointment (Puralube Ointment, Fougera, Melville, NY) was applied topically prior to pricking the vein with the lancet. Approximately 4 to 5 drops of whole blood (equivalent to 100 µl) were collected in a 1.3-ml lithium-heparin tube to prevent clotting. The volume of whole blood relative to volume of anticoagulant was less than recommended; however, the yield of blood that could be obtained from each animal was consistent across all samples. Hemostasis was achieved by using sterile gauze and paper material applied with gentle manual pressure, and animals were returned to their home cages and observed briefly. Blood samples from rats with glomerulosclerosis were collected under ketamine (55 to 65 mg/kg intraperitoneally) and diazepam (6.0 to 6.5 mg/kg intraperitoneally) anesthesia at the conclusion of experimental procedures, prior to perfusion. Blood was allowed to flow directly from the site of cannulation of the aorta, posterior to the kidneys, into lithium-heparin collection tubes.

Figure 1. Equipment needed for collection and processing of whole-blood samples: a lithium heparin (green-topped) 1.3-ml tube, a disposable plastic pipette, the E6+ Mini-Urgent Care cartridge, the i-STAT portable clinical analyzer, and an infrared printer. The standard-size office key is provided for scale.

Anticoagulated blood was drawn into sterile disposable pipettes and expressed into the wells of the cartridges within 30 min of collection, in accordance with manufacturer instructions. Cartridges were stored at 4 °C and warmed to room temperature for at least 5 min prior to application of sample. Quality control is performed automatically by an internal mechanism, wherein the disposable microfabricated cartridge is responsible

Table 1. Analyte intervals for mice (n = 131) based on results from a portable clinical analyzer

Analyte	Published range	Measured range (± 2 standard deviations)	Analyzer mean	% overlap[a]
Glucose (mg/dl)	90–192	89.3–176.2	132.8	85
BUN (mg/dl)	18–29	13.6–34.8	24.2	100
Sodium (mmol/l)	124–174	136.0–148.2	142.1	24
Potassium (mmol/l)	4.6–8.0	4.9–9.0	7.0	91
Chloride (mmol/l)	92–120	109.6–127.7	118.7	37
Hematocrit (%)	35.1–45.4	36.8–52.7	44.8	83
Hemoglobin (g/dl)	11.0–15.2	12.8–17.7	15.3	57

Blood was collected from the submandibular vein.
[a]Percentage of published interval contained in estimated interval.

Table 2. Analyte intervals for rats (n = 76) based on results from a portable clinical analyzer

Analyte	Published range	Measured range (± 2 standard deviations)	Analyzer mean	% overlap[a]
Glucose (mg/dl)	50–135	97.2–157.9	127.6	44
BUN (mg/dl)	9–21	8.5–22.7	15.6	100
Sodium (mmol/l)	135–146	126.3–142.1	134.2	65
Potassium (mmol/l)	4–5.9	4.4–7.7	6.1	79
Chloride (mmol/l)	96–107	99.8–116.0	107.9	65
Hematocrit (%)	34–53	29.5–58.3	43.9	100
Hemoglobin (g/dl)	10–16.7	10.0–19.8	14.9	100

Blood was collected from the lateral saphenous vein.
[a]Percentage of published interval contained in estimated interval.

for all analysis measurements relative to internal standards.[24] Cartridges were sealed by closing the attached cap over the well prior to insertion into the analyzer. Within 2 min, results for the tests were computed, and hard copies were obtained by the infrared transmission of information to the system printer. After use, the E6+ cartridge was removed and discarded.

Published reference values. Glucose, blood urea nitrogen, sodium, potassium, chloride, hematocrit, and hemoglobin levels were recorded, and ranges compared with a compendium of published rodent blood values.[2,4,7] The published ranges for hematocrit and hemoglobin of mice were taken from animals of various ages and of genetic backgrounds that included ICR, BALB/c, C57BL/6, CD1 (ICR), and SW.[7] Information regarding the site of blood collection in mice was not available. The published ranges for hematocrit and hemoglobin for rats were taken from animals of various ages and of genetic backgrounds that included F344, LE, and SD.[7] For these values, blood was collected from the hearts of ether-anesthetized rats. Published, yet provisional, ranges for glucose and blood urea nitrogen (BUN)[4] and electrolytes[2] were provided on routine handouts distributed by the Animal Diagnostic Laboratory at the University of Michigan. Information was not available regarding the specific analyzers used to compute any of these published values. For the purposes of this study, all published ranges provided by the contract lab merely served as target intervals against which to compare analyzer values from laboratory mice and rats.

Statistical methods. Analysis was performed using R statistical software (Department of Statistics and Mathematics, Vienna School for Economics, Vienna, Austria).[5] The mean and standard deviation of the sample values were calculated. The range was defined as the mean ± 2 standard deviations; this definition was based on the assumption of normal distribution among sample test values. This approach generated normal ranges that included approximately 95% of the tested clinically normal animals. Overlap between the published and analyzer ranges was computed as the percentage of the published range that was contained within the ranges established using the analyzer. From both ranges, we computed the difference between the high and low endpoints of the portable clinical analyzer range divided by the difference between the high and low endpoints of the published range, multiplied by a factor of 100% to obtain a percentage value.

Results

When used with mice, the portable clinical analyzer yielded comparable values to those previously published using other analytical methods (Table 1), with 5 of 7 tested analytes showing over 57% overlap with published ranges. Analytes with a greater overall range (that is, lower and upper limits of the analyzer range were outside of the published range) were potassium, chloride, hematocrit, and hemoglobin. The overall range for sodium was narrower than the published range (136.0 to 148.2 mmol/l versus 124.0 to 174.0 mmol/l), with only 24% overlap. The range for BUN values was more broad (13.6 to 34.8 mg/dl versus 18.0 to 29.0 mg/dl), allowing for a 100% overlap with published values. Despite the higher ranges of 4 of the tested analytes, the analyzer ranges were tighter (upper value minus lower value) for glucose, sodium, and chloride, as compared with published intervals.

When used in rats, the portable clinical analyzer yielded values comparable to those previously published based on other analytical methods (Table 2), with 6 of 7 tested analytes showing over 65% overlap with published ranges. The estimated ranges for glucose, potassium, and chloride were higher overall, whereas the interval for sodium was shifted downward. The range for glucose (97.2 to 157.9 mg/dl versus 50.0 to 135.0 mg/dl) had the least overlap (44%) with the published range, yet was also tighter than the published range. Estimated intervals for hematocrit, hemoglobin, and BUN were more broad, with 100% overlap with the published ranges. Comparison of values that fell at least 20% above or below the computed bounds from all mice or all rats revealed no significant differences among

Table 3. Comparison of BALB/c, C3H, and 129/Sv mice using established analyzer ranges for tested analytes

Analyte	Measured range	BALB/c (n = 44)	C3H (n = 49)	129/Sv (n = 38)
Glucose (mg/dl)	89.3–176.2	93.6–174.1	99.9–182.2	83.5–152.1
BUN (mg/dl)	13.6–34.8	**10.7**–34.8	15.6–30.3	**16.7**–37.5
Sodium (mmol/l)	136.0–148.2	132.7–148.1	137.7–147.7	137.0–148.4
Potassium (mmol/l)	4.9–9.0	5.2–8.9	**6.1**–9.1	4.2–8.4
Chloride (mmol/l)	109.6–127.7	112.7–133.0	112.5–124.9	110.0–123.0
Hematocrit (%)	36.8–52.7	35.2–51.9	35.4–50.7	41.7–52.6
Hemoglobin (g/dl)	12.8–17.7	12.0–17.6	12.9–16.7	14.2–17.9

Blood was collected from the submandibular vein.
Values in boldface are more than 20% above or below the corresponding lower limit of the established range for the portable clinical analyzer.

Table 4. Comparison of SD and NIH rats using established analyzer ranges for tested analytes

Analyte	Measured range	SD (n = 50)	NIH (n = 26)
Glucose (mg/dl)	97.2–157.9	109.2–154.0	82.4–156.8
BUN (mg/dl)	8.5–22.7	8.6–22.0	8.4–23.9
Sodium (mmol/l)	126.3–142.1	126.6–140.7	126.1–144.3
Potassium (mmol/l)	4.4–7.7	4.4–7.6	4.2–7.9
Chloride (mmol/l)	99.8–116.0	99.7–116.0	99.8–116.2
Hematocrit (%)	29.5–58.3	27.5–54.7	**40.6**–57.9
Hemoglobin (g/dl)	10.0–19.8	9.3–18.6	**13.8**–19.7

Blood was collected from the lateral saphenous vein.
Values in boldface are more than 20% above or below the corresponding lower limit of the established range for the portable analyzer.

mice and rats of varying genetic backgrounds (Tables 3 and 4). Although these noted differences are not a traditional measure of significance as calculated according to a *P* value, they represent possible phenotypic nuances that may exist for differing genetic backgrounds of rodents or may be due to experimental influences.

A mouse model of type I diabetes and a rat model of glomerulosclerosis were used to assess the ability of the portable clinical analyzer to confirm the biologic effects of these diseases (hyperglycemia and elevated BUN levels, respectively). The diabetic mice were tested under nonfasted and fasted conditions (Table 5). The laboratory previously had confirmed hyperglycemia (assessed as a glucose level greater than 250 mg/dl) in these animals by analysis of whole blood using a human glucometer. Similarly, the values for blood glucose were greatly elevated above the range established using the portable clinical analyzer (539 to greater than 700 mg/dl for fasted mice and 644 to greater than 700 mg/dl for nonfasted mice). In the rats, renal dysfunction had been confirmed by laboratory personnel through the measurement of elevated urine protein:creatinine (greater than 1.5). Our results (Table 6) showed elevated levels for BUN (28 to 136 mg/dl), consistent with kidney dysfunction, whereas BUN values for the control animal fell within the established interval.

Discussion

Our study is the first to report analyte ranges for a substantial number of clinically normal mice (n = 131) and rats (n = 76) by use of a portable clinical analyzer. We compared published rodent values for glucose, BUN, hematocrit, hemoglobin, sodium, potassium and chloride, provided by a contract laboratory, to ranges determined through the use of a portable analyzer. After determining normal ranges, we assessed 2 rodent models of induced disease and verified that expected values for certain analytes (that is, blood glucose for mice and BUN values for rats) fell outside the established analyzer intervals.

Few studies have been published on the evaluation of portable clinical analyzers in laboratory rodents. In one study, pH and partial and total CO_2 in Crl:CD(SD)BR anesthetized rats (n = 20) were evaluated using blood collected from the caudal vena cava.[8] Calcium in rats (n = 16) was measured in blood that was collected from the heart and then pooled and mixed with ethanol prior to assay.[12] In a related study, rats (n = 22) were injected with ethanol and blood was sampled via tail nick for calcium measurements.[13] The analytical cartridge that we used in our study does not contain pH, CO_2 , or calcium in its portfolio; therefore we cannot compare these studies to our described work.

A portable analyzer was used in mice infected with *Candida albicans* to identify renal failure, hyponatremia, hypochloremia, and metabolic alkalosis as contributing components of poor health.[19] The study objective was to validate the pathophysiologic model of human disease by euthanizing groups of animals (n = 8) at specified time points after infection, with blood collected at necropsy after intraperitoneal injection with anticoagulant. Those investigators used a cartridge that included the parameters we used, as well as pH, partial CO_2, bicarbonate, and base excess; an additional cartridge was used to measure creatinine. Renal failure in the mice was determined by elevations in both BUN and creatinine levels, with progressive and simultaneous drops in serum sodium and chloride levels.[19] Although this previous work did not rigorously establish baseline test intervals for a large number of animals, the findings support the use of the same portable clinical analyzer for assessing rodent models of human disease.

More recently, values obtained with a portable analyzer for mice (n = 28) were compared with values obtained from 3 automated clinical chemistry analyzers. The authors did not recommend the interchangeable use of the portable clinical analyzer and other large-scale analyzers within the same study; however, this conclusion was influenced by differences in sample preparation, including the type of anticoagulant

Table 5. Assessment of hyperglycemia in a mouse model of type I diabetes mellitus

Mouse no.	Condition	Glucose (mg/dl)	BUN (mg/dl)	Sodium (mmol/l)	Potassium (mmol/l)	Chloride (mmol/l)	Hematocrit (%)	Hemoglobin (g/dl)
		89.3–176.2	13.6–34.8	136.0–148.2	4.9–9.0	109.6–127.7	36.8–52.7	12.8–17.7
1	Nonfasted	644	21	132	8.7	110	47	16
	Fasted	539	22	139	5.9	109	50	17
2	Nonfasted	>700	53	137	5.9	107	45	15.3
	Fasted	>700	25	133	5.4	99	43	14.6
3	Nonfasted	>700	38	140	6.5	113	45	15.3
	Fasted	586	25	133	6.2	111	41	13.9
4	Nonfasted	>700	39	139	6.3	109	48	16.3
	Fasted	619	18	140	5.1	102	52	17.7

Blood was collected from the submandibular vein.
The range established for each analyte by using the portable analyzer is given at the top of the column.

Table 6. Assessment of impaired renal function in a rat model of glomerulosclerosis

Rat no.	Glucose (mg/dl)	BUN (mg/dl)	Sodium (mmol/l)	Potassium (mmol/l)	Chloride (mmol/l)	Hematocrit (%)	Hemoglobin (g/dl)
	97.2–157.9	8.5–22.7	126.3–142.1	4.4–7.7	99.8–116.0	29.5–58.3	10.0–19.8
1	223	28	132	3.1	114	34	11.6
2	145	47	131	4.4	113	30	10.2
3	82	108	143	3.8	117	25	8.5
4	169	46	137	3.3	113	32	10.9
5	184	36	131	3.7	108	36	12.2
6	199	70	134	2.8	110	28	9.5
7	157	57	136	5.1	113	36	12.2
8	202	136	128	5.8	106	27	9.2
9	172	103	120	>9.0	115	36	12.2
10[a]	219	21	143	2.5	105	46	15.6

Blood was collected by aortic cannulation of rats anesthetized with ketamine and diazepam.
The range established for each analyte by using the portable analyzer is given at the top of the column.
[a]Control animal, injected intraperitoneally with saline.

used and the assessment of differing blood components (whole blood versus serum) using differing analytical methods. The authors of the cited study concluded that although the observed differences between instruments were statistically significant, these differences may not be clinically relevant.[20] Our ranges for all parameters measured in mice (within 95% confidence) were more broad than those reported by others.[20] This result is expected, given the greater number of animals that were enrolled in our study. The published result most discordant with our work was the greater range of normal glucose (209 to 241 mg/dl). This difference from our findings might be attributed to the intracardiac method of blood collection after CO_2 inhalation, as both of these factors may lead to the stress response of transient hyperglycemia. We used blood collection techniques that typically do not require anesthesia to avoid its potential influence on reported values.

Before using an analyzer for experimental studies on laboratory rodents, reference ranges for the instrument must be established using appropriate sample sizes (at least 47 to 97 animals) that assure inclusion of a clinically normal population within a 90% to 95% confidence interval.[20,23] Our established ranges can be used both to assess disease phenotypes and to evaluate individual rodents that present with a variety of clinical aberrations (for example, dermatitis, diarrhea, lethargy). Typically mice and rats are not assessed as individual patients; however, numerous clinical conditions in rodents can be monitored and managed for improved individual, as well as colony, health. Most rodent strains and experimental models represent a considerable investment of research effort and cost toward specific medical model development. Therefore, we encourage the use of portable clinical analyzers as an option for antemortem clinical assessments prior to postmortem diagnostics.

We experienced minimal problems with the procurement of test results. An error report occurred if the cartridge was not preloaded with the minimal sample volume. In some cases, potassium values exceeded the quantitative range of the instrument (greater than 9.0 mmol/l), although the mice were clinically normal. These values may have represented individual variability for certain animals or artificial elevations due to sample clotting. Hyperkalemia was measured in mice of all 3 strains; however, the number of animals affected (n = 7) was low, and we included their analyte panels in our analysis. Potassium, along with glucose and phosphorus, has been reported to be unstable in samples of whole blood;[16] therefore we processed all samples within 30 min of collection.

In the confirmation of hyperglycemia and elevated BUN in the experimental rodent models, some measured analytes fell outside of our established ranges; this could be related to the physiologic impact of the induced diseases. In addition, variations in glucose and electrolyte levels for the control rat (no. 10)

could be related to handling, injection of saline or anesthetic, or underlying subclinical abnormalities. Our intent in this study was not to rigorously evaluate these 2 disease models; the small sample sizes of experimental animals were selected only to determine whether the portable clinical analyzer could verify disease in affected rodents through a testing mechanism other than that used currently by each investigative group. Future work will focus on phenotyping rodent models of induced disease with serial evaluations of clinical disease development.

Continued demand for laboratory mice and rats as biomedical models of disease necessitates the refinement of diagnostics for these species. Making veterinary medical decisions for these species requires the establishment of baseline reference intervals for the species of interest on the analyzer that will be used. Handheld clinical analyzers, which are portable, easy to use, and require little maintenance, have the capacity for multiple clinical applications in the specialty field of laboratory animal medicine.

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