Measurement of urinary glycosaminoglycans in dogs

David C. Grant, DVM, MS; S. Dru Forrester, DVM, MS; David L. Panciera, DVM, MS; J. Blair Meldrum, DVM, PhD

Objectives—To measure urine concentrations of sulfated glycosaminoglycans (GAGs), determine optimal storage conditions for urine samples, establish a reference range, and determine whether there is correlation between 24-hour total urine GAG excretion and the GAG-to-creatinine ratio (GCR).

Animals—14 healthy adult dogs.

Procedure—Single urine sample GAG concentrations and GCRs were measured in samples collected from 14 healthy dogs at the start of the 24-hour collection period. Twenty-four-hour total urine GAG excretions were determined from urine collected during a 24-hour period in the same 14 dogs. Total sulfated GAG concentrations were also measured in urine from these dogs after the urine had been stored at 4°C and –20°C for 1, 7, and 30 days.

Results—Urine GAG concentrations were not significantly different from baseline values after urine was stored at 4°C for up to 1 day and –20°C for up to 30 days. Neither single urine sample GAG concentration (R², 0.422) nor GCR (R², 0.084) was an adequate predictor of 24-hour total urine GAG excretion.

Conclusions and Clinical Relevance—Results of this study provide data that can be used to establish a reference range for 24-hour total urine GAG excretion in dogs and adequate conditions for sample storage. Contrary to findings in humans, there was no significant linear correlation between 24-hour total urine GAG excretion and single urine sample GCR in dogs, limiting clinical use of the single urine sample test. (Am J Vet Res 2006;67:51–55)

Heparan sulfate, dermatan sulfate, keratan sulfate, and chondroitin sulfate are types of sulfated GAG chains found in human urine.³ Measurement of urinary excretion of GAGs in human and veterinary medicine has been used to evaluate the pathogenesis of various diseases and treatments, including mucopolysaccharidoses, rheumatoid arthritis, urolithiasis, neoplasia of the urinary tract, extracorporeal shock wave lithotripsy, diabetes mellitus, interstitial cystitis, renal failure, glomerulonephritis, and renal amyloidosis.²⁻¹⁴ Of particular interest are altered GAG excretion in human patients with glomerular diseases.²,¹¹,¹³,¹⁵,¹⁶ There is a paucity of reports¹⁸ on urine GAG measurements in dogs.

The urine GCR for single urine samples differed significantly between clinically normal humans and patients with primary glomerulonephritis or 2 forms of renal amyloidosis.⁵,¹⁶ In those studies, urine GCRs in humans with glomerulonephritis and amyloidosis were significantly decreased, compared with values for clinically normal humans. Analysis of these results suggests that the 1,9-DMB spectrophotometric assay of urine sulfated GAGs has the ability to distinguish clinically normal humans from those with primary glomerulonephritis and amyloidosis. The authors of those studies believed this test may also be able to differentiate between patients with glomerulonephritis and those with amyloidosis. In contrast, another study¹ conducted by other investigators found an increase in urine GCR in humans with glomerulonephritis. Differences in GCR in these studies²,¹⁵,¹⁶ appear to be attributable to variation in preparation of buffer solutions, differences in the absorption wavelengths, questionable statistical analysis, and possible interfering substances.

The purpose of the study reported here was to determine whether the DMB spectrophotometric method could be used to accurately quantify concentrations of sulfated GAGs in urine samples obtained from dogs. Additionally, correlation of the single urine sample GCR with 24-hour total urine GAG excretion and methods of sample preservation were determined for urine samples obtained from clinically normal dogs.

Materials and Methods

Animals—Fourteen random-source adult dogs were used in the study. Dogs were considered healthy on the basis of results of physical examination, a CBC, serum biochemical analysis, ELISA to detect infection with Dirofilaria immitis, urinalysis, urine protein-to-urine creatinine ratio, and aerobic bacteriologic culture of urine samples. Dogs were fed a commercially available food formulated for dogs until 12 hours before the start of collection of urine samples. Dogs were allowed ad libitum access to water until the start of the collection of urine samples. The procedures in this study were approved by the Virginia Tech Animal Care Committee.

Procedure—Urine samples were collected from each dog for a period of 24 hours to determine the normal 24-hour total urine GAG excretion. Dogs were administered acepro-

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From the Departments of Small Animal Clinical Sciences (Grant, Forrester, Panciera) and Biomedical Sciences and Pathobiology (Meldrum), Virginia-Maryland Regional College of Veterinary Medicine, Virginia Tech, Blacksburg, VA 24061. Dr. Forrester's present address is Hill's Pet Nutrition Inc, PO Box 148, Topeka, KS 66601.
Supported by a grant from the Veterinary Memorial Fund.
Address correspondence to Dr. Grant.

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mazine maleate (0.01 to 0.02 mg/kg, IM). Approximately 15
20 minutes later, each dog was weighed and then admin-
istered water (volume equivalent to 5% of body weight) via
an orogastric tube to approximate daily maintenance water
requirements and ensure hydration during the collection
period. Neither food nor water was provided during the 24-
hour collection period to prevent contamination of urine in
the collection pans.

A urethral catheter was inserted into the urinary bladder
of each dog, and the bladder was emptied at the beginning of
the 24-hour collection period. Single urine sample creatinine
and GAG concentrations were measured in this initial urine
sample; analysis was conducted within 4 hours after collect-
ion of the initial urine sample. At the start of the 24-hour
collection period, 3 mL of blood was collected from each dog
and used to determine baseline (time 0) plasma concentra-
tions of glucose, urea nitrogen, creatinine, phosphorus,
calcium, total protein, albumin, globulin, total bilirubin, cho-
lesterol, sodium, potassium, chloride, and total carbon dio-
X
400

Assay of GAG concentration —The procedure used to
determine sulfated GAG concentrations in canine urine was
adapted from a technique used in humans. This method was
chosen because it is not affected by proteinuria, a problem
detected with other versions of the DMB-based GAG assay.
Chondroitin-4-sulfate was used as a representative sulfated
GAG to approximate total sulfated GAGs. Briefly, a stock
solution (10.0 mg/mL) of C-4-SO₄ in deionized water was
prepared. The same stock solution was used for all tests.
Aliquots of stock solution were placed in separate test tubes
and diluted appropriately with deionized water to achieve
concentrations of 0, 0.25, 0.5, 0.75, 1.0, 1.5, 2.0, 3.0, 4.0, 5.0, 7.5, and
10.0 mg/mL for use in establishing an initial standard curve.
A stock solution of DMB was prepared by adding 11 mg
of DMB to 1 L of 0.05M sodium acetate buffer (pH 4.75). This
stock DMB solution was stored at 23°C in a light-protected
brown glass bottle with an airtight cap. Additional stock solu-
tion of DMB was prepared as needed and was stored for no
longer than 1 month. No precipitate was observed at any time.
All urine samples were processed by centrifugation at 400 × g for 5 minutes. Supernatant was harvested and used for
analysis. An aliquot (250 µL) of supernatant was added to
2.5 mL of DMB solution in each cuvette, the cuvette was
inverted twice, and change in absorbance was measured
within 1 minute. Change in absorbance caused by each urine
sample was measured in triplicate at 520 nm, which was one
of the absorbance peaks of the DMB–C-4-SO₄ complex.
A spectrophotometer was used for determination of
change in absorbance of C-4-SO₄ stock solutions and urine
samples. Disposable cuvettes with a 1-cm length of light path
and volume of 4.5 mL were used. Change in absorbance of
2.5 mL of the stock solution of DMB (with deionized water
used as a control sample) was determined in triplicate at 350,
400, 450, 500, 520, 535, 550, 600, 650, and 700 nm each day
to ensure stability of the dye solution.

Each day that urine was assayed, a standard curve (C-4-
SO₄ concentrations of 1, 2, 4, 7.5, and 10 mg/dL) was creat-
ed, and simple linear regression analysis was performed to
determine the relationship between change in absorption and
C-4-SO₄ concentration. To determine whether there was a
change in absorbance over time, C-4-SO₄ concentrations of
0.0, 0.25, 1.0, 4.0, and 10 mg/dL were measured in triplicate at
1, 2, 3, 4, 5, 10, 20, and 30 minutes. No obvious change in
absorbance was detected.

Regression lines for the sulfated GAG standard curves
were assessed for linearity, and an R² value was calculated by
use of commercially available software. An R² > 0.98 was
considered strong evidence of a linear relationship, and the
corresponding regression equations were used to calculate
sulfated GAG concentration from the change in absorbance
of each urine sample.

Measurement of creatinine concentration —Urine sam-
ple was centrifuged at 400 × g for 5 minutes. Supernatant
was harvested and used for measurement of the creatinine
concentration. Creatinine concentration was determined by
an automated chemistry analyzer by use of the modified Jaffé
procedure.

Endogenous Ccreat was calculated by use of the following
equation:

\[ C_{\text{creat}} = \frac{(U_{\text{creat}} \times V)}{(PC \times X \times BW)} \]

where Ucreat is the urine creatinine concentration, V is
the volume of urine, PC is the mean plasma creatinine con-
centration in samples obtained at baseline and at the end of
the urine collection period, T is the time of the collection period,
and BW is body weight.

Statistical analysis —Data from all dogs that completed
the 24-hour urine collection were used to study effects of
urine storage on GAG concentrations. Only data from dogs
with a Ccreat > 2 mL/min/kg were used for determination
de GAG concentrations and GFR, 24-hour total urine GAG excretion, and correlations among these
variables.

The GAG concentrations of the stored urine samples were
analyzed by use of a complete randomized block design to
detect effects of time and temperature. Difference between
the GAG concentration at time 0 and at each time and temperature
combination was the variable used for analysis. A repeated-
measures ANOVA was performed by use of commercially
available statistical software. Values of P < 0.05 were consid-
ered to indicate significant differences. Simple linear regression
was used to determine the line of best fit for the relationship of
24-hour total urine GAG excretion with single urine sample

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GCR and GAG concentration. The $R^2$ values were calculated by use of commercially available software as an estimate of the relationship among these variables.

Results

All 14 dogs completed the 24-hour urine collection period. The urine GAG concentration did not differ significantly from baseline values when samples were stored at 4°C or –20°C for 1 day or at –20°C for 30 days, but concentrations were significantly different at all other time and temperature combinations (Table 1). Urine GAG concentrations changed in a similar manner for both storage temperatures.

Of the 14 dogs that completed the 24-hour urine collection, 10 had a $C_{\text{creat}} > 2$ mL/min/kg (mean ± SD, 2.581 ± 0.352 mL/kg/min). This group consisted of 6 spayed females, 3 sexually intact males, and 1 neutered male. Mean body weight was 21.57 kg. The age of each dog was unknown, but all were adults. Data from the 4 dogs with a $C_{\text{creat}} < 2$ mL/min/kg were excluded from determination of values for clinically normal dogs.

For the 10 dogs with a $C_{\text{creat}} > 2$ mL/min/kg, mean ± SD 24-hour total urine GAG excretion was 1.586 ± 0.461 mg/kg/d. Mean GCR for the 24-hour total urine sample was 0.044 ± 0.012. Mean single urine sample GAG concentration and GCR were 5.007 ± 1.588 mg/dL and 0.023 ± 0.01, respectively. No significant linear relationship could be detected between single urine sample GAG concentration and 24-hour total urine GAG excretion or single urine sample GCR and 24-hour total urine GAG excretion (Figures 1 and 2).

Discussion

Urine GAG concentrations remained stable when urine samples were stored at 4°C or –20°C for 1 day.
magnitude to values reported for humans deter-
ple GAG concentration and GCR in the 10 dogs com-
excretion. In 1 study, investigators found that
dehydration or approximates 24-hour total urine GAG
corrects the GAG concentration for variable states of
conflicting information in humans as to whether GCR
to the urine creatinine concentration. However, there is
fluctuation throughout the day. Nearly all reports in
resulted if excretion of GAGs or creatinine were not
are unknown. Lack of correlation also could have
interaction tubing. Effects of dander, hair, and interactions
was in contact with the stainless-steel cage and collec-
samples may have resulted because urine for the latter
excretion among animals. If single urine sample GCR
cannot be used, a 24-hour urine collection would be
necessary, which is impractical for routine use in vet-
erinary medicine. Additionally, this would require an
indwelling catheter, and the effects of an indwelling
catheter on GAG concentration are unknown.

Several possible reasons exist for lack of a correla-
tion between single urine sample GCR and 24-hour
total urine GAG excretion. Physical differences
between the single urine samples and 24-hour urine
samples may have resulted because urine for the latter
was in contact with the stainless-steel cage and collection
tubing. Effects of dander, hair, and interactions
with the tubing on GAG and creatinine concentrations
are unknown. Lack of correlation also could have resulted
if excretion of GAGs or creatinine were not constant,
even if there is nonparallelism in their fluctuation throughout the day. Nearly all reports in
which investigators have measured urine GAG concen-
trations have expressed the results as a ratio in relation
to the urine creatinine concentration. However, there is
conflicting information in humans as to whether GCR
corrects the GAG concentration for variable states of
dehydration or approximates 24-hour total urine GAG
excretion. In 1 study, investigators found that
GAG excretion measured in single, random urine
samples had good linear correlation with 24-hour total
urine GAG excretion. In another study, investigators
used the same GAG assay that we used in the study
reported here and found daily and circadian urine
excretions of GAG were constant in 5 humans. However, when urine GAG concentrations are esti-
ated by measuring hexuronic acid or uronic acid
concentrations, there may be nonparallelism in daily fluc-
tuation of GAG and creatinine concentrations. The
GCR measured in a single random sample may fluctu-
ate throughout the day, and there may also be day-to-
day fluctuations, but the GCR for 24-hour urine sam-
ple remains consistent from day to day. In the study
reported here, the single urine sample GCR was mea-
sured in a sample collected from each dog in the morn-
ing and was lower than the GCR for the total 24-hour
sample in all 10 dogs (Figure 3). This implies that excretion of GAGs or creatinine is not constant in
dogs. Differential effects of hydration status on pro-
duction, excretion, and reabsorption of creatinine and
GAGs should be considered, but have not been report-
ed in humans or dogs to our knowledge.

Water gavage was used in place of ad libitum con-
sumption of water to ensure hydration and prevent
contamination of the collected urine. However, this
gavage did add the variable of forced diuresis and may
have altered GAG excretion during the subsequent col-
lection period. Additional studies involving ad libitum
access to water and collection of single, random urine
samples multiple times throughout a 24-hour period
may help clarify any contribution hydration status and
time of sample collection may have had on our find-
ungs. Such studies may also reveal conditions for which
the single urine sample GCR is a viable measurement
for clinical studies.

The study reported here provided data that can be
used to establish a reference range for 24-hour total
urine GAG excretion in dogs. At this point, it must be
concluded that single urine sample GCR is not an ade-
quate predictor of 24-hour total urine GAG excretion.
On the basis of analysis of results of our study, use of
24-hour urine collections would be needed to make
comparisons among specific animals. Additionally, this
information should cause clinicians and researchers to
use caution when interpreting results of experiments
in which single urine sample GCR is used to compare
urine GAG excretion in humans and in cats with inter-
stitial cystitis.

a. Lab-Line multiwrist shaker, Lab-Line Instrument Inc, Melrose
Park, Ill.
b. DU640B spectrophotometer, Beckman Coulter, Fullerton, Calif.
c. Microsoft Excel 2000 software, Microsoft Corp, Redmond, Wash.
d. Olympus AU 400 automated chemistry analyzer, Olympus
America Inc, Diagnostic Systems Group, Melville, NY.
e. SAS System software, version 8.2, SAS Institute Inc, Cary, NC.

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