mens from D–IV levels, eggs of the phylum Acanthocephala were found (Figs. 1 and 2) (*8*). All eggs were remarkably similar in form and size, and showed identical staining properties by iodine. No other species were identified.

Members of the phylum Acanthocephala are unique in several respects (*9*). The phylum consists of one class and three orders, is exclusively parasitic, and utilizes birds, fish, and mammals as definitive hosts. Only two species, *Macracanthorhynchus hirudinaceous* and *Moniliformis dubius*, within the order Archiacanthocephala, have been implicated in human disease, and both are cosmopolitan in distribution. *Macracanthorhynchus hirudinaceous*, or giant thorny-headed worm, is a helminth parasite of swine (peccary) and, occasionally, of the dog and monkey. It was reported once as occurring in a human in 1859 on postmortem examination of a 9-year-old child in Prague (*12*). In the life cycle of *Macracanthorhynchus hirudinaceous*, the egg is ingested by scarabaeid beetles or their grub. The beetles serve as the intermediate host where the infective larval stage develops.

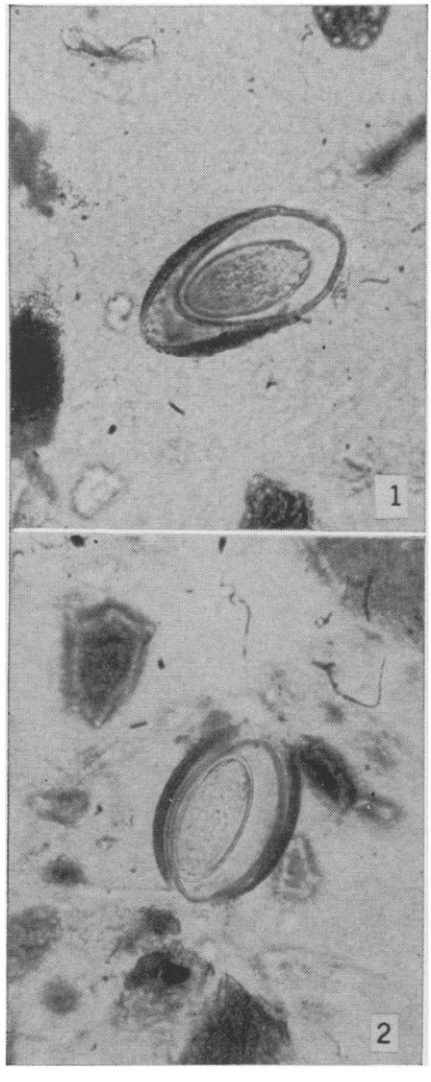

Fig. 1. Acanthocephala egg emerging from protective shell found in coprolite from Danger Cave level IV (1869 B.C. ± 160 years). The egg measures 73 by 44 μ (dimensions between outer membranes excluding the shell). Fig. 2. Egg within thick shell found in D–IV coprolite. The egg measures 72 by 46 μ (dimensions between outer membranes excluding shell).

The definitive host of *Moniliformis dubius* is primarily the rat; beetles and cockroaches serve as intermediate hosts. Human infestations have been reported from Sudan, Italy, and British Honduras (*12*). In Utah near Danger Cave, the closely related form *Moniliformis clarki* occurs today (*10*). The intermediate host is the camel cricket *Ceuthophilus utahensis*, and definitive hosts include a variety of small rodents (*11*). Other insects probably act as intermediate hosts and the adult form probably occurs in other mammals, although examination of coyotes in the area has not revealed infestation (*11*). Species differentiation is not possible from the morphological characteristics of the eggs; still, it is likely that the eggs from the Danger Cave coprolites are *Moniliformis clarki*.

Infestation in man by *Moniliformis clarki* has not been reported. Nevertheless, a few inferences may be drawn. Historically, aboriginal peoples commonly ate insects, and no doubt prehistoric man did the same. Larval grub forms, crickets and grasshoppers (*13*), and probably beetles and cockroaches were an integral part of the diet. Small animals, including rodents, are known to have been eaten whole (*13*). Thus, aboriginal people could have served as a definitive host by ingesting the arthropod intermediate host, or they may have been victims of false parasitism as a result of eating parasitized rodents. Since the definitive host for *Moniliformis clarki* is not specific, a large natural host reservior existed in a variety of vertebrates in the vicinity of Danger Cave, including man.

In what manner Acanthocephala infection affected community health and individual life expectancy remains speculative. The worm is armed with a formidable proboscis which burrows into the intestinal wall causing diarrhea, weight loss, anemia, emaciation, and, not uncommonly, death from perforation in the host (*9*). In the only recorded experimental infection in man with *Moniliformis dubius* (1888), symptoms of severe abdominal pain, diarrhea, exhaustion, somnolence, and tinnitus were produced 19 days after ingesting several larvae (*12*). Cure was effected by administration of male fern extract (*Aspidium filixmas*) within 2 days. Thus, unlike parasitism by the more common helminths—*Ascaris lumbricoides, Enterobius vermicularis*, or *Trichuris trichiura*—Acanthocephala infection probably affected individual health and had lethal potentialities.

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References and Notes

1. F. O. Callen, *New Sci.* **8**, 35 (1960).
2. R. F. Heizer, *Univ. Calif. Archaeol. Surv. No. 70* (April 1967), p. 1.
3. G. F. Fry, thesis, Department of Anthropology, University of Utah (1968).
4. A. W. Pike, *Disease in Antiquity* (Thomas, Springfield, Ill., 1967), chap. 14.
5. R. Samuels, *Amer. Antiquity* **31**, 175 (1965).
6. H. J. Van Cleave and J. A. Ross, *Science* **105**, 139 (1947).
7. J. D. Jennings, *Univ. Utah Anthropol. Pap. No. 2* (1957), p. 1.
8. Identification by M. Chitwood, Beltsville Parasitological Laboratory, USDA, Beltsville, Md.
9. A. U. Jones, *Introduction to Parasitology* (Addison-Wesley, Reading, Mass., 1967), chap. 12.
10. J. R. Crook and A. W. Grundman, *J. Parasitol.* **50**, 689 (1964).
11. A. W. Grundman, personal communication.
12. E. C. Faust and P. F. Lussell, *Clinical Parasitology* (Lea & Febiger, Philadelphia, ed. 6, 1957), chap. 32.
13. J. H. Steward, *Univ. Calif. Publ. Amer. Archaeol. Ethnol.* **33**, 255 (1933).

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Glass-Transition Temperature of Water

Abstract. *The glass-transition temperature of water* (T_g) *has been calculated by use of the Tammann-Hesse viscosity equation with the viscosity equal to* 10^{13} *poise at* T_g. *The derived value of* T_g, *162 ± 1°K, is significantly higher than previous estimates.*

Yannas (*1*) has commented on the importance of the glass-transition temperature T_g of water in meteorological and other investigations; he reported an estimate of this value based on volumetric measurements of glycerol-water solutions. I here report an alternate

Table 1. Viscosity parameters for water.

log A	B (°K)	T_0 (°K)
−3.54	222	149.4 (*5*)
−3.54	224	148.4 (*6*)
−3.58	226	149.4 (*6*)
−3.54	222	150 (*4*)

method for deriving T_g, which gives an appreciably higher value.

The viscosities of many liquids over broad temperature ranges can be accurately represented by the equation (*2*)

$$\log \eta = \log A + B/(T-T_0) \quad (1)$$

where η is the viscosity at temperature T, and A, B, and T_0 are constants. The extrapolation by this equation of the viscosities of glass-forming liquids to T_g, as determined by volumetric or calorimetric measurements, gives log $\eta_g = 13 \pm 1$ (*2*, *3*). For example, for glycerol (*2*), log $A = -4.76$, $B = 950$, and $T_0 = 132$°K. Substitution of these values in Eq. 1, with log $\eta_g = 13$, gives $T_g = 185$°K, the correct value as cited by Yannas.

The viscosity parameters for water have been determined in several independent investigations giving essentially the same values (Table 1).

Using Eq. 1, with log $\eta_g = 13$, we obtain $T_g = 162° \pm 1$°K, which is appreciably higher than $127° \pm 4$°K, the value derived by Yannas. This result suggests that T_g for glycerol-water solutions might go through a minimum somewhere above a water weight-fraction of 0.4, the upper limit of Yannas' measurements. If we consider the correlation between viscosity and structure due to H-bonding which has been demonstrated for pure water (*4*), a possible minimum in the constant-viscosity temperature T_g has interesting implications regarding the structural variation as a function of composition in supercooled glycerol-water solutions.

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References

1. I. Yannas, *Science* **160**, 298 (1968).
2. G. Tammann and W. Hesse, *Z. Anorg. Allgem. Chem.* **156**, 245 (1926).
3. A. J. Barlow, J. Lamb, A. J. Matheson, P. R. K. L. Padmini, J. Richter, *Proc. Roy. Soc. London Ser. A. Math. Phys. Sci.* **298**, 467 (1967); A. A. Miller, *J. Polymer Sci. Part A-2* **6**, 1161 (1968).
4. A. A. Miller, *J. Chem. Phys.* **38**, 1568 (1963).
5. F. Guttmann and L. M. Simmons, *J. Appl. Phys.* **23**, 977 (1952).
6. M. Hoffmann and K. Rother, *Rheol. Acta* **2**, 164 (1962).

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Teleostean Urophysis: Stimulation of Water Movement across the Bladder of the Toad Bufo marinus

Abstract. *An effect of material from the caudal neurosecretory system of a teleost on the isolated toad bladder is described. Urophysial breis from* Gillichthys mirabilis *result in dose-related water movement across the bladder. As little as one one-hundredth of a urophysis induces a threefold increase in osmotic water movement.*

The caudal neurosecretory system of fishes and its neurohemal organ in teleosts, the urophysis, form an endocrine apparatus whose function has proven remarkably elusive (*1*, *2*). Recently a water-retaining effect in toads was reported (*3*). The isolated toad urinary bladder was examined to determine whether it played a role in this water-retention effect and whether the degree of response was dose-dependent and could therefore be used in the development of an assay. Previous reports had indicated little or no hydrosmotic influence of urophysial material, as can be seen with various neurohypophysial peptides (*2*, *4*). The present report describes a pronounced, dose-related response of the toad bladder to urophysial preparations.

Table 1. Effects of 0.1 of a urophysis (UH) from *Gillichthys* per milliliter and equivalent amounts of abdominal spinal cord (SC) on water loss in milligrams by isolated toad bladders.

First period	Second period	Third period	Fourth period	Fifth period
10.3	143.7 (UH)	4.5	5.3 (SC)	4.9
6.8	128.2 (UH)	4.9	4.0 (SC)	4.1
10.3	21.5 (SC)	6.6	152.2 (UH)	5.1
8.5	8.8 (SC)	5.4	122.8 (UH)	4.4

The toads (*Bufo marinus*) were obtained from Hawaii and kept in the laboratory at 22°C. The procedure for the preparation of toad bladder as described by Bentley (*5*) was followed with certain modifications. A hemibladder, tied to the end of a glass tubing, was filled with 1 ml of an amphibian Ringer solution (*5*) (*p*H 8.2 to 8.3), immersed in 30 ml of the same solution, and allowed to equilibrate for 3 to 4 hours. For the last hour of equilibration, the inside solution was changed to 20 percent Ringer solution, and the outside solution was replaced by fresh Ringer. Before weighing, the bladder was suspended inside a balance and immersed in Ringer in a funnel (also inside the balance) connected to a reservoir by rubber tubing. When the bladder was to be weighed, the Ringer solution was drained from the funnel by lowering the reservoir. This setup has the advantage of leaving a constant amount of water adhering to the surface of the preparation at the time of weighing.

The bladder preparation was weighed before and after 30-minute periods to determine water loss. Urophysial and other materials were added to the external solution 10 minutes before each test period; intervening 30-minute periods served as controls. Each hemibladder was subjected to a maximum of three experimental periods alternated with four control periods. After each 30-minute period, there was an interval of about 30 minutes during which the bladders were rinsed by refilling and placing in fresh solution. During all these periods and during rinsing, the bladder contained 1 ml of 20 percent Ringer solution and was immersed in 30 ml of undiluted Ringer solution at room temperature.

Urophyses from mudsuckers (*Gillichthys mirabilis*) were homogenized in Ringer solution with a glass homogenizer. Whole homogenates were used throughout. *Gillichthys*, a Pacific Coast goby (20 to 30 g) is rather easily collected in tidepools and is a hardy euryhaline fish.

A good dose-response relation was obtained with three doses of the urophysial preparations, given in increasing or decreasing order (Fig. 1). As little as 0.01 of a urophysis, equivalent to about 0.2 μg of acetone-dried urophysis powder (*6*), per milliliter of Ringer solution increased water loss to about three times the control values. Doses of 0.03 and 0.1 of a urophysis per milliliter resulted in increases of approximately 8- and 16-