The Auk 109(3):451–464, 1992

ONTOGENESIS OF INTESTINAL NUTRIENT TRANSPORT IN DOMESTIC CHICKENS (*GALLUS GALLUS*) AND ITS RELATION TO GROWTH

BRYAN S. OBST[1] AND JARED DIAMOND

Department of Physiology, University of California Medical School, Los Angeles, California 90024, USA

ABSTRACT.—We measured intestinal (brush-border) uptake of the sugars glucose and fructose, and of the amino acid proline, each week from one day posthatching to 12 weeks of age in a rapidly growing strain of domestic chicken (*Gallus gallus*, White Cornish × Rock). Small-intestine mass increased as the 0.74 power of body mass, the same power as resting metabolic rate of the whole bird. Even on a constant diet, nutrient uptake varied with age, according to ontogenetic patterns that differed among nutrients. From the beginning to end of the 12-week period, glucose uptake per milligram of intestinal tissue remained on the average approximately constant, fructose uptake increased in the distal intestine, and proline uptake per milligram declined across all intestinal regions. Uptake capacity integrated over the whole length of the intestine generally increased for all three solutes, mostly as a consequence of increases in intestinal mass. A steep, brief, many-fold increase in glucose uptake appeared in two-week-old chicks, coinciding with increasing thermal independence and the exhaustion of yolk reserves. A similarly steep, brief, many-fold increase in proline uptake at week 6 coincided with the onset of molt and high relative growth rates. Comparisons of integrated nutrient-uptake capacities with estimated nutrient intakes suggest that intestinal nutrient uptake capacity is closely matched to dietary nutrient intake during growth. This supports the hypothesis that the intestine's capacity to assimilate nutrients may pose a proximal constraint on the rate of growth in precocial species. *Received 1 November 1990, accepted 6 February 1992.*

THE FACTORS that determine growth rates among birds have provoked interest for decades. As a group, birds include the fastest-growing vertebrates (Case 1978); yet, birds exhibit an impressive array of species-specific growth rates that vary with adult body size, degree of precociality, and ecology (Lack 1968, Ricklefs 1968, 1976, 1983). Because mortality is high in vulnerable young birds, many authors have argued that natural selection should favor maximum-possible rates of growth (Ricklefs 1969, Martin 1987). While early studies emphasized food limitations as the ultimate source of constraint (Lack 1968, Case 1978), some recent studies have searched for more proximal physiological factors that might constrain growth rates below values that food availability would permit. These studies have usually emphasized limitations set by ontogenetic programs influencing differential rates of organ development and maturation (Ricklefs 1979, 1983, Lilja 1983).

Recent studies on the ontogenesis of sugar and amino-acid uptake in the intestines of several mammal species suggest that intestinal uptake capacity is closely matched to intake during postnatal growth (Buddington and Diamond 1989, 1990, Toloza and Diamond 1992). These findings raise the possibility that intestinal absorption may impose limits or "bottlenecks" to the availability of nutrients, which in turn would constrain rates of growth. Among birds, patterns of intestinal growth appear to be correlated with body growth rates (i.e. rapidly growing birds exhibit rapid early development of digestive organs; Lilja 1983). Such observations have led to the hypothesis that growth rate may be determined in part by the relative allocation of tissue to the alimentary tract (Lilja et al. 1985, Konarzewski et al. 1989).

In order to evaluate whether intestinal nutrient uptake presents a bottleneck to avian growth, we used the everted-sleeve technique (Karasov and Diamond 1983) to measure rates of intestinal brush-border uptake for two sugars (glucose, fructose) and one amino acid (proline) during postembryonic development in the domestic chicken (*Gallus gallus*). Briefly, the intestine is lined by a single layer of absorptive cells termed enterocytes, whose cytoplasm is sepa-

[1] Deceased.

rated from the intestinal lumen and from the bloodstream by the so-called brush-border membrane and the basolateral membrane, respectively. Intestinal nutrient absorption involves at least four major steps in sequence: hydrolysis of high-molecular-weight nutrients, such as proteins and starch, by soluble enzymes (such as pancreatic amylase); hydrolysis of resulting medium-molecular-weight nutrients, such as peptides and small disaccharides, by enzymes bound to the brush-border membrane (such as sucrase); uptake of resulting low-molecular-weight nutrients, such as simple sugars and amino acids, into the enterocyte by transport proteins in the brush-border membrane; and export of those nutrients to the bloodstream by transport proteins in the basolateral membrane. In the steady state, time-averaged net fluxes for a given class of nutrient must be equal through these sequential steps, after correction for factors such as nutrient metabolism, synthesis, or loss to the lower intestine. There is no evidence that any one of these four steps is rate-limiting compared to the others; instead, activities of the enzymes and transporters at all four steps are regulated by dietary nutrient levels (Diamond and Hammond 1992).

We focus on the step of brush-border uptake, which is effected by carrier proteins in the brush-border membranes (transporters) specific to each class of nutrient. Many transporters consume energy supplied through the sodium concentration gradient. The sleeve of intestinal tissue is everted in order to expose the enterocytes, which normally face inward toward the lumen of the intestine, to the oxygenated bathing solution *in vitro*. Although we measure uptake *in vitro*, the everted sleeve method yields uptake rates comparable to *in vivo* values measured by modern methods (Toloza and Diamond 1992).

We selected *Gallus* for several reasons. First, the nutritional requirements for growth are better known in the domestic chicken than in any other avian species. Second, *Gallus* chicks are self-feeding from hatching; chicks can be raised on a diet of constant composition, and food intake rates are determined by the chick rather than by the parents. This is a major advantage: diet composition changes with age in all growing mammals and many growing birds, making it difficult to separate the relative contributions of diet changes and of genetic programs to observed changes in intestinal nutrient uptake. Finally, the heavy-bodied strain that we selected (White Cornish × Rock cross) is the product of centuries of intense artificial selection for rapid and efficient growth; thus, physiological adaptations and constraints to rapid growth may be particularly emphasized in this strain. In fact, the unutilized excess capacity by which intestinal uptake capacity exceeds nutrient intake in growing mammal species proves to be virtually absent in the chicken strain that we studied.

Although there have been several previous studies of the development of intestinal transport in chickens (Bogner and Haines 1964, Holdsworth and Wilson 1967, Shehata et al. 1981, Lerner et al. 1976, Raheja et al. 1977, Planas et al. 1982, 1986), each has focused on one or two solutes, one or two intestinal regions, and a restricted span of ages. The present paper represents the most comprehensive study of the ontogenesis of intestinal nutrient transport in a bird, and the first attempt to link developmental patterns of intestinal uptake with functional aspects of avian growth.

METHODS

Animals and their care.—Male White Cornish × Rock cross chicks ($n = 48$) were delivered on their day of hatching to the UCLA Center for Health Sciences vivarium, where they were raised in indoor cages. Chicks were given constant (24-h) light and were provided *ad libitum* water and food (Universal Feeds all-purpose mash; analysis, 18.0% crude protein, 2.2% crude fat, crude fiber <13%, ash <15%) throughout the study. Chicks were first housed in groups of 12 to 15 in commercial brooders. After four weeks, the growing chicks were transferred to larger stainless-steel cages (~1 m^3), where they were housed in pairs or groups of three for the remainder of the study. Food and water were replaced at least twice daily to ensure freshness and constancy of supply. Chicks were inspected weekly for signs of damage or disease; seven chicks with damaged feet and stunted growth were discarded during the study.

Resting metabolic rate.—Rates of oxygen consumption (VO_2) were measured using a Beckman E-2 paramagnetic oxygen analyzer in a flow-through respirometry system. Each week from day 0 to week 12 two or three individuals (total $n = 29$) were isolated in darkened cages without food or water for 6 h. At the end of that time each bird was weighed and transferred to a darkened metabolic chamber maintained at a temperature (26–28°C) within chicks' thermal-neutral zone (Misson 1982) and equivalent to temperatures inside the aviaries. Air, dried and scrubbed of CO_2, was passed at a flow rate of 500 to 2,000 cc/min to the oxygen analyzer. Gas volumes were corrected to units of standard temperature and pressure,

and VO_2 was calculated by equation 2 of Hill (1972). Measurements did not begin until the bird had been in the chamber for at least 1 h and, clearly, had settled down following the transfer.

We defined the resting metabolic rate (RMR) of a given individual as the lowest VO_2 maintained for at least two 5-min periods during an hour of measurement. RMR is not strictly synonymous with basal metabolic rate (BMR), since the chicks were probably not fully postabsorptive (Nir and Nitsan 1979) and since their metabolism includes a component associated with growth. However, RMR is otherwise similar to BMR in that it represents the metabolic rate of a bird at rest, in the dark, and in thermal neutrality; therefore, it is a measure of the minimum rate of oxidative metabolism in a normally growing bird.

Nutrient-uptake measurements.—The everted-sleeve technique has been previously described in detail (Karasov and Diamond 1983) and adapted to birds (Karasov et al. 1986a, Obst and Diamond 1989); it will be outlined only briefly here. Three or four chicks (distinct from the individuals used in respirometry studies) were removed from the cages at one-week intervals through 12 weeks. The chicks were anesthetized with intraperitoneal injections of Nembutal. The abdomen was opened, and the entire gut (small intestine, ceca, colon) was rinsed out with an ice-cold avian Ringer's solution (recipe in Obst and Diamond 1989) and excised. The intestine was divided into several sections according to the anatomical definitions of McClelland (1979:132–141): three regions of the small intestine (duodenum, jejunum, and ileum), the paired ceca, and the colon. The length of each region was measured and the region was then everted over a glass rod to expose its luminal surface. A number of 1-cm segments of "sleeves" were taken from the middle of each region and mounted on glass rods of appropriate diameter (2–10 mm, chosen to yield a snug fit to the sleeve) using silk ligatures separated by 1 cm. The mounted sleeves were stored in ice-cold, oxygenated Ringer's solution for up to 1 h before transport was measured.

Each mounted sleeve was pre-incubated for 5 min in avian Ringer under physiological conditions of pH (7.4), temperature (40°C), and oxygenation (95% O_2, 5% CO_2). After this period, the sleeve was transferred to an incubation solution containing the nutrient of interest (D-glucose, D-fructose, or L-proline). The aldohexose D-glucose and the ketohexose D-fructose move on different transporters, which together represent the major pathways of hexose sugar uptake in vertebrates. L-proline has its own "private" transporter, but also uses three other amino-acid transport pathways (Lerner 1984, Karasov et al. 1986b, Obst and Diamond unpubl. data) and, therefore, measures the sum of four of the five important pathways of amino-acid uptake. Each rod-mounted sleeve was incubated for 2 min, a period within the linear range of uptake versus incubation time for both glucose and proline. Temperature, pH, and oxygenation were as described for the pre-incubation, but the incubation solution was also stirred at 1,200 rpm with a magnetic stir bar to reduce the effects of unstirred layers on uptake. Concentration of the sugar or amino acid was 50 mM, and the total osmolality in each incubation solution was 350 mOsm (300 mOsm inorganic salts plus 50 mM of nutrient). A nutrient concentration of 50 mM was selected on the basis of measurements of uptake as a function of concentration, indicating that this concentration saturated or nearly saturated the respective carriers for D-glucose, D-fructose, and L-proline.

Rates of uptake were determined from accumulation of radiolabeled solutes in the tissues. Each incubation solution contained two labeled compounds: the nutrient of interest (i.e. ^{14}C-labeled D-glucose and D-fructose, ^{3}H-labeled L-proline) and an extracellular space marker with the complementary label (^{3}H L-glucose for the sugars, ^{14}C polyethylene glycol [PEG] for proline). For proline, the PEG allowed us to correct the total quantity of the amino acid present in the sleeve for the quantity that was present in the adherent fluid and extracellular space but not absorbed by the epithelium. (PEG is too large a molecule to diffuse through the epithelium.) For D-glucose and D-fructose incubations, L-glucose allowed correction for both the sugar in the extracellular space and the sugar accumulated in the sleeve by passive permeation. (L-glucose has the same diffusion coefficient as D-glucose or D-fructose, but is not subject to carrier-mediated transport.) Thus, measured uptake of D-glucose and D-fructose represents only the carrier-mediated component, while proline uptake includes both carrier-mediated and passive diffusional components. Validation experiments show that the sodium-independent fraction of total proline transport in chick jejunum is 45%. However, since proline moves on both sodium-dependent and sodium-independent carriers (Lerner 1984), the passive (noncarrier-mediated) component is less than 45%.

After incubation, sleeves were daubed at their tips with Kimwipes to remove excess adherent fluid, removed from the rods, and placed in preweighed scintillation vials. Vials were reweighed to determine wet masses of each sleeve to the nearest 0.1 mg. Sleeves were digested in tissue solubilizer, followed by addition of scintillation cocktail, and the activities of ^{3}H and ^{14}C were determined via liquid scintillation counting to an accuracy of $\pm 0.1\%$. Tissue uptakes were calculated using equations provided in Karasov and Diamond (1983).

Integrated-uptake calculations.—To compare small-intestine uptake capacity to nutrient demands, we combined measurements of uptake per centimeter length of individual sleeves with measured lengths of the intestine in order to estimate the integrated uptake capacity of the whole small intestine for each nutrient. Regional uptakes per sleeve (uptake/cm) were

multiplied by the length of each intestinal region (cm/region), and these products (uptake/region) were summed for the duodenum, jejunum, and ileum to yield integrated uptake (uptake/intestine). In this manner, we were able to calculate an integrated uptake capacity for each nutrient in each individual chick. Although we also measured uptake in the paired ceca and colon, uptake capacities in these regions of chicks were slight (Obst and Diamond 1989), accounting for less than 3% of integrated uptake for all three solutes we studied. Therefore, we chose to systematically exclude cecal and colonic uptake values from our calculations of integrated uptake, and they are not further discussed in this paper.

Our comparison of integrated uptake capacity to nutrient intake/demand involves several assumptions and approximations (Buddington and Diamond 1990, Ferraris et al. 1990). First, carrier-mediated uptake rates are saturable functions of concentration, and we measured uptake at concentrations (50 mM) chosen to yield nearly maximal uptake rates; one cannot automatically assume that actual luminal substrate concentrations in the chick will also yield maximal uptake rates. We know of no measurements of luminal concentrations in chickens, but several lines of evidence suggest that this assumption is not unreasonable. Chicks were given unlimited access to food on a schedule of 24-h light, and they fed both day and night. Nir and Nitsan (1979) demonstrated that chicks seldom completely empty their guts, but draw gradually upon crop fill even when offered food only intermittently. Ferraris et al. (1990) demonstrated that luminal concentrations for glucose are typically near or above the K_m (concentration of half-maximal carrier saturation) in the intestinal chyme of rats, rabbits, and dogs. Hence, we expect uptake levels prevailing *in vivo* to be within a factor of 2 of the maximum rates of uptake that we measured *in vitro* at 50-mM concentrations.

Second, in order to relate integrated uptake to metabolic demand, our measurements should encompass all the important pathways of macronutrient uptake. We selected glucose as a marker for carbohydrate uptake because it is the most abundant dietary sugar (from the hydrolysis of starches). A separate, sodium-independent transporter exists for fructose, although it is a less important component of grain. Digestive hydrolysis of proteins yields 20 common amino acids, which are then absorbed as monomers by the intestinal brush border. Amino-acid absorption is very complex in chickens, involving at least five separate transporters with broad, overlapping specificities for the 20 common amino acids (Lerner 1984). As already mentioned, proline has access to at least four of these carriers and, therefore, proline uptake measures the sum of most of the capacities for amino-acid uptake in general. Proline is excluded from one important carrier specific to acidic amino acids (Lerner and Steinke 1977), and our measurements of integrated uptake therefore neglect transport for this class of non-essential (but abundant) amino acids. Lipids move passively across the intestinal wall and are also excluded from our measurements. However, fats provided less than 3% of the dry mass and roughly 8% of calories in the experimental diet, and nearly all the energy for the growing chick was satisfied by dietary carbohydrates and proteins.

Metabolic equivalents.—In order to compare measurements of metabolizable energy intake, oxygen consumption, and intestinal nutrient transport, it was necessary to express these in common units. VO_2 was converted to units of power (W or kJ/day), assuming an energy equivalence of 20.1 kJ per L of O_2 consumed. Units of integrated uptake (nmoles/intestine,min) were converted to power assuming an energy equivalence of 16.7 kJ/g for sugar and 18.8 kJ/g protein. Values for metabolizable energy intake (kJ/week) were taken from published values for broiler-breed chicks (National Research Council 1984) with similar diets and growth rates as our study population. Metabolizable energy was partioned into fractions attributable to carbohydrate (60.3%), protein (31.1%), and fat (8.6%) based on their relative proportions in the diet analysis and energy equivalents listed above.

Statistics.—Values in the text are given as the mean ± SE. Vertical bars in the figures span one SE above and below the mean. Differences between means were tested for significance using Student's two-tailed *t*-test or ANOVA (Sokal and Rohlf 1981), and a significance level of $P < 0.05$.

RESULTS

Body growth.—Chicks increased more than 50-fold in body mass from 57.1 ± 4.0 g ($n = 6$) one day posthatching to over 3,000 g in 12 weeks (Fig. 1). Although chicks continued to increase in mass beyond this age (~4,200 g at seven months), the first 12 weeks represent the phase of highest body growth. Absolute growth rate was highest in the period between seven and nine weeks of age, when on the average 386 g/week were added. Relative growth was fastest in the first week posthatching (100%/week) and declined generally, though not monotonically, through the twelfth week of life.

Resting metabolic rate.—RMR increased with body mass over the first 12 weeks. This increase was allometric (Fig. 2), as described by the equation:

$$\mathrm{RMR} = 0.073 M_B^{0.80 \pm 0.03}, \quad (1)$$

where RMR is $cm^3\ O_2/min$ and M_B is body mass

in grams. RMR was highly mass-dependent; body mass alone explained 98.7% of the variance in RMR.

Intestinal growth.—The length of the small intestine increased by a factor of 3.45, from 53.7 ± 3.0 cm (n = 4) after hatching to 185.5 ± 3.6 cm (n = 4) at 12 weeks (Fig. 3). However, because the intestine thickened as well as lengthened with age, the mass of the entire small intestine increased 15-fold over the same period (Fig. 3). Intestinal growth was allometric (Fig. 2), as described by the equation:

$$M_I = 0.237 M_B^{0.74 \pm 0.06}, \quad (2)$$

where M_I is intestine mass in grams and M_B is body mass in grams. This exponent did not differ significantly from the allometric exponent (0.80 ± 0.03) relating RMR to body mass (P > 0.25). Hence, intestinal mass remains directly proportional to metabolic mass, even though relative intestinal mass declines with age. Relative intestinal growth was especially fast in the first week of life: intestine mass tripled while body mass doubled. At week 2 the small intestine accounted for an average of 9.4 ± 2.3% of the total body mass. By week 12, this percentage had declined to only 2.5 ± 0.2%.

Although all three regions of the small intestine increased in mass with age, rates of change were different for each region. Gut length increased more rapidly in the jejunum and ileum than in the duodenum, while unit mass (mg/cm) increased more rapidly in the duodenum and jejunum than in the ileum (Fig. 3). As a result, the mass of each region exhibited a separate allometry, with all three regions starting at roughly the same mass in hatchlings, but the jejunum increasing its contribution to total intestinal mass throughout development (Fig. 3).

Regional nutrient uptake.—Normalized to wet-tissue mass, rates of uptake for glucose, for fructose, and for proline did not vary widely between small-intestine regions over the first 12 weeks of development (Fig. 4). There was, however, a consistent tendency for glucose uptake to be highest in the jejunum throughout development (significant at hatching and weeks 3, 6, 10, and 11; t-test, P < 0.05) and for proline uptake to be lowest in the duodenum (P < 0.05 at weeks 1, 4, and 7).

Tissue-specific uptake of glucose (nmoles/min mg) was similar for chicks just after hatching

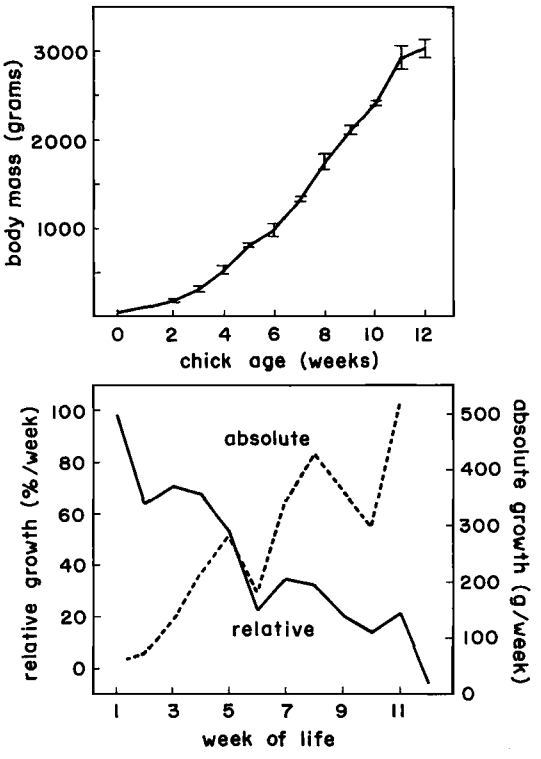

Fig. 1. Postembryonic growth of *G. gallus* chicks during first 12 weeks after hatching. In this and subsequent figures, vertical bars span 1 SE above and below the mean.

and at 12 weeks of age in all three regions of the small intestine (P > 0.10 for all three pairs of means). However, a pronounced spike in tissue-specific glucose uptake occurred in chicks at two weeks of age, when uptake increased by factors of 5.0, 6.1, and 8.8 in the duodenum, jejunum and ileum, respectively. In the jejunum and ileum, glucose uptake reached a mean of 31.6 ± 9.7 nmoles/mg,min. This increase was transitory, and uptake levels returned to their previous levels by week 3.

Tissue-specific uptake of fructose exhibited little net change over the course of development in the duodenum or jejunum, and exhibited rather wide variation between individuals. Fructose uptake was consistently lower than glucose uptake in all ages in all intestinal regions. However, there was a trend for fructose uptake to increase steadily with age in the ileum, where specific fructose uptake more than tripled, from 0.43 ± 0.12 nmoles/mg,min just after hatching to 1.39 ± 0.28 nmoles/mg,min

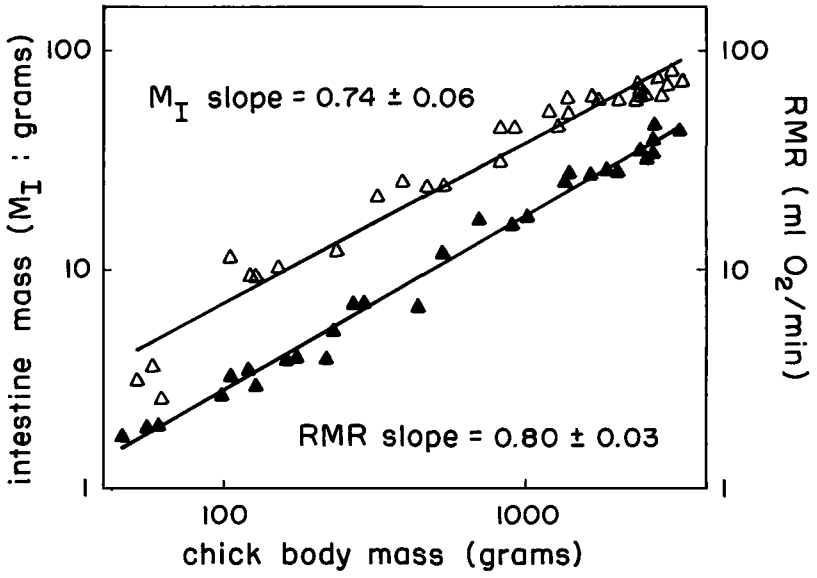

Fig. 2. Allometric changes in resting metabolic rate (RMR, closed symbols) and small-intestine wet mass (M_I, open symbols) during development in domestic chicken. Note that slopes (± 95% confidence intervals) of least-square regression lines relating log RMR and log M_I to log body mass are not statistically different ($P > 0.25$).

at week 12 ($P < 0.05$). Unlike the case for glucose, there was no surge in fructose uptake at week 2.

Proline uptake showed a general decline with age, especially evident over the first four weeks posthatching. This decline was consistent in all three regions of the small intestine. As with glucose uptake, the general ontogenetic trend was interrupted by a transitory surge in uptake; however, with proline this surge occurred at week 6, four weeks later than the glucose spike. Tissue-specific proline uptake increased by factors of 3.1, 4.3, and 2.3 between weeks 5 and 6 in the duodenum, jejunum, and ileum, respectively. Mean proline uptake rates at week 7 were statistically indistinguishable from those measured at week 5, but both were significantly lower than values measured at week 6 ($P < 0.05$).

Integrated nutrient uptake.—The intestine's integrated uptake capacity is the product (summed over intestinal length) of tissue-specific uptake rates and of intestinal quantity, both of which changed with age. Not surprisingly, integrated uptake for each of the three nutrients generally increased with age and with the increasing size of the intestine (Fig. 5). However, the pattern of increase was not monotonic for either glucose or for proline, owing primarily to the transitory spikes in tissue-specific uptake described above for these nutrients (Fig. 4). The jejunum contributed most to integrated uptake, but this was less marked for proline than for sugars.

Table 1 compares integrated uptake with estimates of nutrient intake. Integrated uptake for sugars (glucose + fructose) closely matched estimated intake of carbohydrate throughout the growth period. On a weekly basis, the uptake: intake ratio ranged from 0.4 to 3.6, averaging 1.07 ± 0.23 across the weeks. Similarly, integrated proline uptake closely matched protein intake (excluding acidic amino acids); uptake: intake ratios ranged between 0.6 and 2.3 and averaged 1.25 ± 0.15.

After converting the rate of uptake to units of power, we can compare the gut's capacity to absorb nutrients with the minimum energy requirements of the growing chick. At all ages, the power equivalent of the intestine's integrated uptake capacity (glucose + fructose + proline) was above the measured resting metabolic rate (Fig. 6). Energy assimilated in excess of RMR is presumably available for activity and growth. In fact, our estimate of integrated uptake roughly approximated metabolizable energy intake over the 12-week period (Fig. 6).

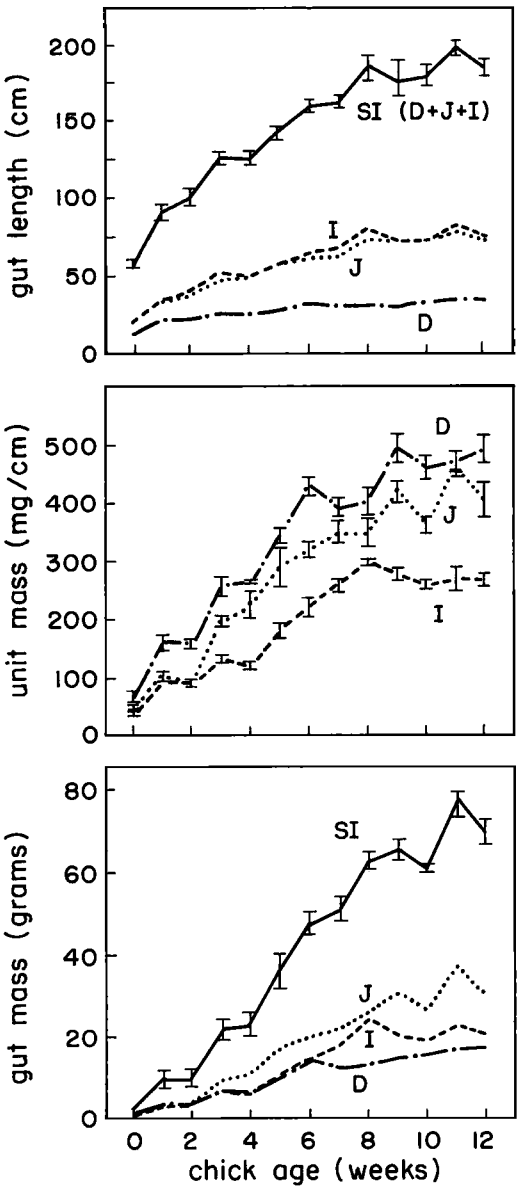

Fig. 3. Growth of the small intestine (SI) during postembryonic development of domestic chicken. Note that both length (cm) and unit mass (mg/cm) of each intestinal region (D = duodenum, J = jejunum, I = ileum) increase during development, but at different rates. As a consequence, the three regions begin with approximately same mass at hatching, but diverge with age.

Thus, even ignoring the less important uptake pathways for other sugars, amino acids, and lipids, there appears to be a close match between the small intestine's uptake capacity and the metabolic needs of the growing animal.

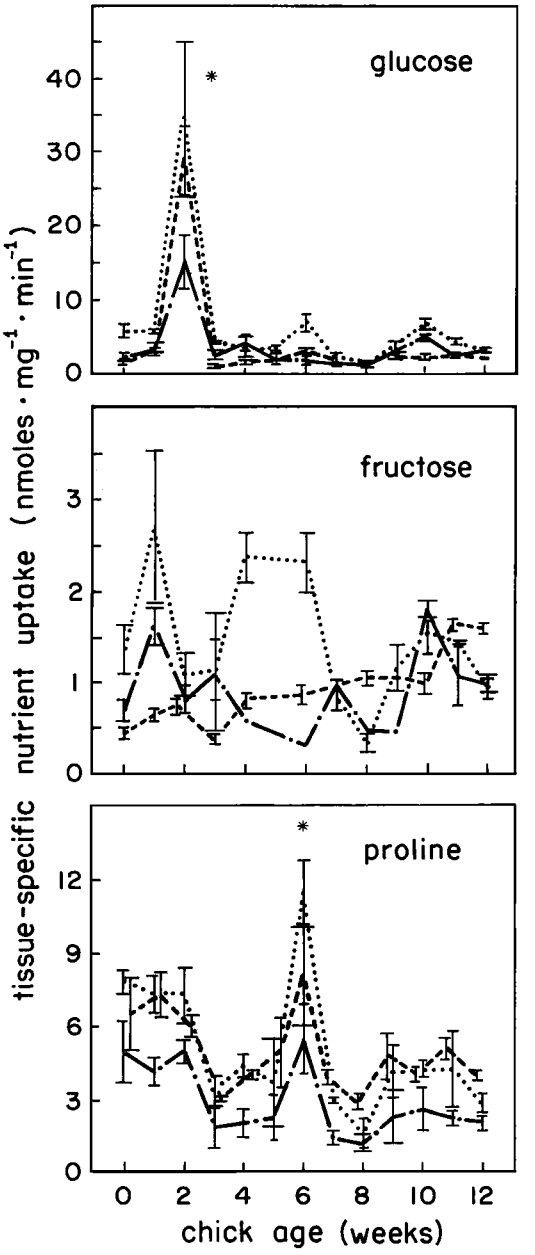

Fig. 4. Rates of uptake normalized to tissue wet mass, for glucose, fructose, and proline measured in everted sleeves taken from three regions of small intestine at various ages. Uptake measured at nutrient concentrations of 50 mM and incubation times of 2 min for all three nutrients. Symbols denoting three regions are as in Figure 3. Asterisks denote weeks during which mean uptake rates were significantly higher than during the immediately preceding and following weeks. Note different ordinate scales for uptakes of three nutrients.

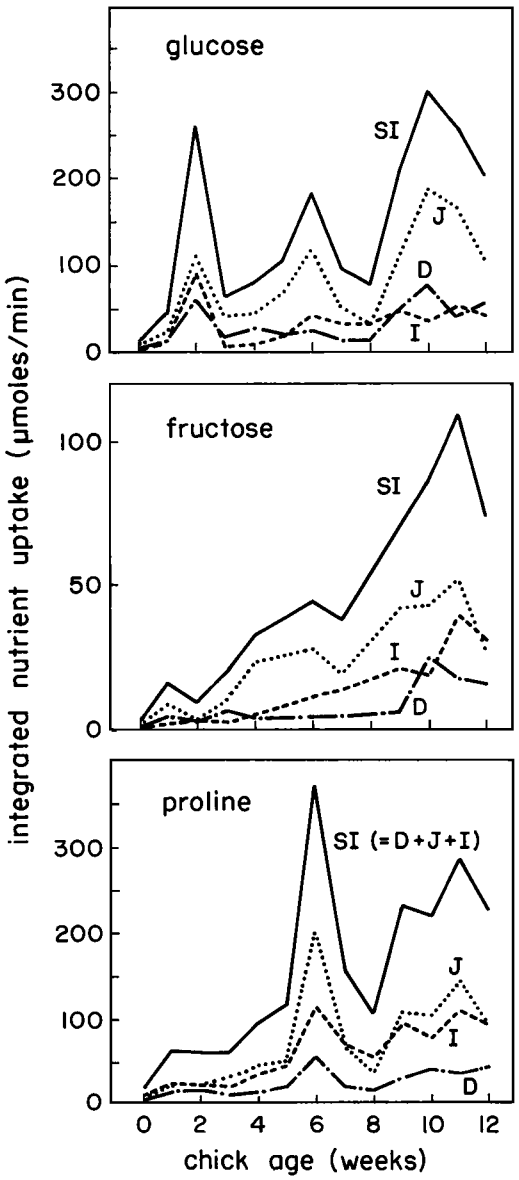

Fig. 5. Integrated uptake of glucose, fructose, and proline as a function of age in domestic chicken. Regional uptakes for the duodenum (D), jejunum (J), and ileum (I) were summed to give total values for small intestine (SI). Note that fructose uptake is drawn to a different scale.

DISCUSSION

The period of postnatal growth is a challenging one for rapidly growing vertebrates. Developmental programs must regulate not only the progressive accretion and maturation of tissues, but also the differential allocation of limited energy and materials to the various functional systems. This challenge is extreme in precocial birds which must locomote, thermoregulate, and forage effectively soon after hatching. Growth rates tend to be lower in precocial birds than in similar-sized altricial birds (Ricklefs 1983, Case 1978), presumably because energy and materials get shunted away from growth and to these competing systems. However, since access to food is not determined by parental feeding, the factors which limit the overall availability of nutrients to precocial chicks are especially interesting.

INTESTINAL GROWTH

Several authors have suggested the possibility that digestive constraints set proximal limits to the rate of avian growth (Lilja 1983, Ricklefs 1983, Konarzewski et al. 1989). Comparative studies (Lilja 1983, Konarzewski et al. 1990) indicate that rapidly growing bird species have rapidly growing guts. Within species, experimental selection for high rates of body growth prove to be associated with increased relative growth of the intestines in lines of Japanese Quail (*Coturnix coturnix*) and domestic chickens (Katanbaf et al. 1988).

In *Gallus*, growth of the intestine is allometric; as the chick grew, a progressively smaller fraction of its total body mass was devoted to intestinal tissue. However, the ontogenetic increase in intestinal quantity (small-intestine mass) kept pace with metabolic rate, because the allometric slopes for intestinal mass and metabolic rate (0.74 vs. 0.80) were not statistically distinguishable ($P > 0.20$). Lilja (1983) reported similar allometric growth coefficients for total gut mass versus body mass in Japanese Quail (0.61) and turkey (*Meleagris gallopavo*; 0.69), and a direct relationship (regression coefficient of 1.0) between intestinal mass and food intake in quail (Lilja 1982). Thus, since the intestine grows in direct proportion to the age-related increase in metabolic rate and food intake, intestinal growth in galliforms plays a fundamental role in meeting the chick's increasing nutrient demands during post-embryonic growth.

ONTOGENESIS OF NUTRIENT TRANSPORT

Using methods identical to ours, other recent studies have investigated the ontogeny of intestinal sugar and amino-acid transport in nu-

TABLE 1. Comparison of intestinal (integrated) uptake for glucose, fructose, and proline with intake of dietary carbohydrates and protein, as a function of age in the domestic chicken.[a]

Age (weeks)	Integrated uptake (kJ/day)			Dietary intake (kJ/day)			Uptake : intake	
	Glucose	Fructose	Proline	Carbo-hydrates	Protein	Protein*	Glucose + fructose (carbo-hydrates)	Proline (protein*)
0	53	11	53	67	49	33	0.95	1.61
1	210	76	196	209	137	92	1.37	2.13
2	1,246	48	195	358	235	157	3.62	1.24
3	298	98	190	508	334	223	0.78	0.72
4	354	154	292	705	460	308	0.72	0.95
5	495	187	376	947	622	417	0.72	0.90
6	864	219	1,163	1,140	749	502	0.95	2.32
7	454	195	497	1,273	832	557	0.51	0.89
8	371	189	367	1,474	968	849	0.38	0.57
9	978	342	755	1,517	996	667	0.87	1.13
10	1,487	422	892	1,551	1,019	683	1.23	1.31
11	1,018	537	931	1,586	1,056	708	0.98	1.31
12	961	365	736	1,638	1,075	721	0.81	1.02
$\bar{x}$							1.07	1.25
SE							0.23	0.15

[a] Dietary intakes calculated by multiplying total metabolizable energy intakes for each age class (National Research Council 1984:table 7) by proportion of energy provided by carbohydrates (60%) and by proteins (31%) in diet. Protein intake adjusted (protein*) by multiplying protein intake by 0.67, the rough proportion of cereal proteins comprised by amino acids other than the acidic amino acids glutamic and aspartic acid (National Research Council 1984:table 25). The reason for this adjustment is that acidic amino-acid uptake is by a carrier not utilized by proline, so that our proline-based measurements of amino-acid uptake capacity do not include capacity for acidic amino acids.

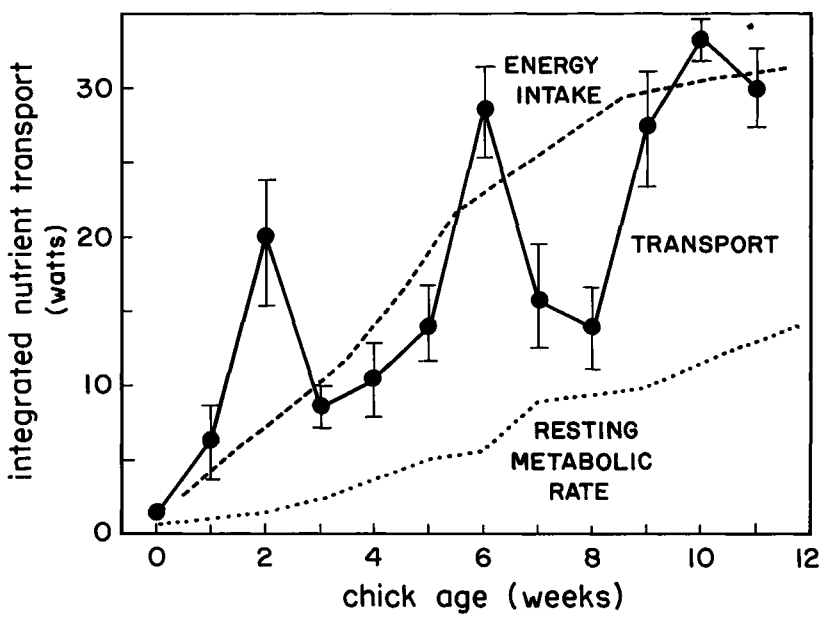

Fig. 6. Comparison of three quantities as function of age in domestic chickens: integrated (whole-intestine) nutrient uptake capacity for proline + glucose + fructose ("transport"); dietary intake of metabolizable energy; and resting metabolic rate. All values expressed in units of power (watts). Note that integrated uptake capacity always exceeds minimum energy demand (RMR) and approximates metabolizable energy intake throughout development.

merous vertebrate species, including fishes (Buddington et al. 1987), amphibians (Toloza and Diamond, 1990), and mammals (Buddington and Diamond 1989, 1990, Toloza and Diamond 1992). The developmental programs of these animals are often characterized by the presence of abrupt ontogenetic events (such as weaning or metamorphosis) accompanied by shifts in diet that complicate the interpretation of intestinal changes. Since diet was constant in our chickens, and feeding rates were determined by the growing animal itself, we can unambiguously ascribe all changes in intestinal uptake in chickens to genotypic programs of development. Nevertheless, our study confirms certain general patterns in ontogenesis of uptake that have emerged from previous vertebrate studies. First, changes in uptake levels for various nutrients occur independently of one another and independently of diet switches. Second, tissue-specific uptake rates need not change monotonically with age ("mature"), but can undergo rapid and reversible regulation. Thus, in chickens as in other vertebrates, great temporal flexibility is evidenced in the developmental program for sugar and amino acid uptake. Finally, chickens share a general trend with many other omnivorous vertebrates for amino-acid uptake to decline and fructose uptake to increase with age, a trend corresponding to the age-related decline in dietary protein and rise in dietary carbohydrate in omnivorous vertebrate species (Buddington and Diamond 1989).

Several aspects of the postnatal development of intestinal transport in chickens stand out as extraordinary. Most striking and unprecedented in any vertebrate species previously studied by our method are the steep and transitory surges in uptake that occur for glucose at week 2 and for proline at week 6. These spikes occur across all regions of the small intestine and result in concomitant surges in integrated uptake capacity. For glucose, both the speed and the magnitude of the change are unparalleled among studies of developing vertebrates. Mean tissue-specific glucose uptake values at week 2 (26.0 ± 6.5 nmoles/mg,min) are comparable to the highest values ever measured in vertebrates (ca. 30 nmoles/mg,min in hummingbirds; Karasov et al. 1986a). Raheja et al. (1977) found a similar (but more muted) pattern in leghorn chicks, where hexose uptake by everted sacs peaked at two weeks.

Proline uptake exhibited a less-marked spike (ca. three-fold increase), but again such a sudden increase is unparalleled in developmental studies. The maximal values reached during this surge (8.4 ± 1.4 nmoles/mg,min) were similar to the mean values we measured in chicks just after hatching (6.5 ± 0.8; t-test for difference between means not significant, $P > 0.25$), and were also comparable to maximum values observed in studies of developing mammals cited above (8–10 nmoles/min,mg). Thus, for the proline spike, the suddenness of the change—but not the absolute rate of transport—is remarkable.

We do not know what cellular mechanism accounts for the developmental changes in tissue-specific uptake rates in *Gallus* chicks. However, rapid turnover of the intestinal epithelium has been reported in young chicks (Imondi and Bird 1966), with minimum cell lifetimes of 48 h from cell division to sloughing of enterocytes in the jejunum. If these lifetimes are correct, they would provide a plausible mechanism for such dramatic changes, since the cell population of the epithelium might easily replace itself entirely between our weekly measurements; each new cell generation provides an opportunity for a change in the expression of transport during its differentiation. This mechanism of adaptation is widespread in adult mammals but contrasts sharply with neonatal mammals, in which enterocytes are believed to have a long life span (Altmann and Enesco 1967, Jarvis et al. 1977, Smith and Jarvis 1978), and in which changes in intestinal transport are attributed to the accretion of cells rather than their turnover. Whatever the mechanism, the remarkable changes observed during development in the chicken warrant a functional explanation.

FUNCTIONAL SIGNIFICANCE OF DEVELOPMENTAL CHANGES

Although changes in nutrient uptake occur continuously over the course of development, we shall consider the functional significance of changes associated with four stages of postnatal development: hatchlings, week 2, week 6, and late development.

Hatchlings.—Day-old chicks exhibit high rates of proline uptake and a high ratio of proline to glucose integrated uptake capacity (P:G ratio; Fig. 7). At hatching, the body cavity is tightly packed with viscera, and intestinal quantity

comprises a smaller proportion of the total body mass than in the two weeks that follow. Nevertheless, relative body growth is maximal during the first week (100%/week), and the corresponding demand for energy and protein must also be high. *Gallus* chicks are precocial but are not fully independent at hatching. Over the first week thermogenetic, thermoregulatory, and locomotory performance improves. In nature, maternal behavior (brooding) compensates by providing protection and energy in the form of shared body heat; in aviaries, chicks huddle together and seek warmth from artificial heaters. Furthermore, maternal nutrients are still present in the hatchling. Lipid-rich yolk reserves provide a parenteral source of energy important to precocial hatchlings (Romanoff 1944, Marcstrom 1966, Peach and Thomas 1986). Thus, we suggest that the hatchling intestine emphasizes amino-acid uptake over sugar uptake because the chick is dependent on dietary protein for rapid growth, but may draw on yolk and fat reserves to obtain calories.

Week 2.—The marked increase in glucose transport and concomitant drop in P:G ratio at this age coincide with several important ontogenetic events. First, chicks gain significant independence, both thermal and locomotory. This independence increases energy expenditures and the need for calories. Secondly, between week 1 and week 2 the yolk reserves become exhausted. Thus, essentially all calories must now be assimilated through the intestine. At the same time, allometric growth of the intestine causes a relative decline in its size (6.4 ± 0.8% of body mass). We suggest that, in order to compensate, the intestine increases uptake of glucose, the nutrient most likely to provide these calories. Body growth continues during this same period, but relative growth (63%/week) is lower than in the first week (100%) or the following week (72%). Hence, while exhaustion of the yolk reserves at a time of high growth rate necessitates the surge in glucose transport in week 2, the subsequent decline in growth rate may contribute to the decline in glucose uptake from week 2 to week 3.

Week 6.—Proline uptake increases dramatically, and the P:G ratio again approaches 2.0, where it remains until week 8 (Fig. 7). This coincides with two ontogenetic events associated with increased protein metabolism: the onset of the first postjuvenile molt (Lucas and Stettenheim 1972); and the rise of absolute rates of

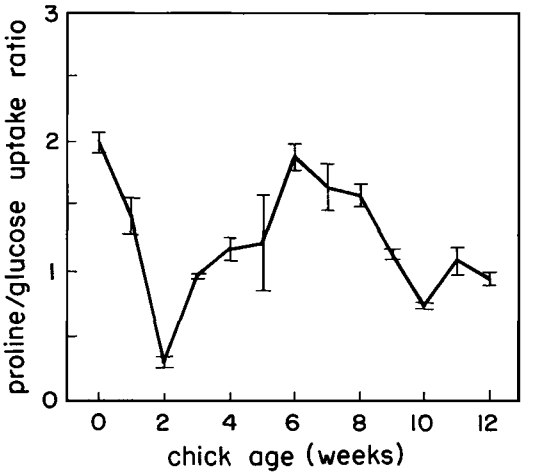

Fig. 7. Ratio of integrated proline uptake capacity to integrated glucose uptake capacity as function of age in domestic chickens. Note extreme values associated with week 0, week 2, and week 6.

body growth. Relative growth rate increased by over 50% (21% per week vs. 33% per week) between weeks 6 and 7. The coincidence of rapid feather and body (muscle) growth during this period should place a premium on protein assimilation. In White Leghorn chickens, Carlotti (1971) reported steep increases in circulating (plasma) levels of free amino acids between about four and eight weeks of age, a period associated with the first molt (Lucas and Stettenheim 1972) and high growth rates (National Research Council 1984). We believe that the increased proline uptake may be part of a more widespread pattern of enhanced amino-acid uptake, transfer, and deposition in the chick.

Late development.—Between weeks 9 and 12, tissue-specific uptakes for both glucose and proline have leveled off. This period is associated with declining relative growth rates of the body (Fig. 1) and intestine (Fig. 3). Although some growth may continue through the following six months, relative growth rates are modest after week 12 (1–2%/week), particularly after males reach sexual maturity (ca. 15 weeks). The most pronounced change in uptake during late development is for fructose, where integrated uptake increases several-fold. As growth rates decline, natural diets might normally be expected to shift away from high-protein foods toward foods containing more glucose and fructose, such as fruits. Dramatic increases in fructose uptake are also seen in rabbits and rats at the

time of weaning, when a similar dietary shift from protein towards carbohydrate occurs, but not in cats, whose normal diet contains little carbohydrate after weaning (Buddington and Diamond 1989, 1990, 1992).

Digestive bottlenecks.—Does the intestine's ability to absorb nutrients actually limit the rate of growth? Integrated over the entire growth period of the chicken, intestinal uptake capacity appears to be closely matched to the chicken's nutrient intake and requirements. Integrated uptake for glucose + fructose + proline is always in excess of resting energy metabolism, but is comparable to total metabolizable energy intake. For sugars and for amino acids, ratios of intestinal uptake capacity to dietary intake average near 1.0. In contrast, uptake:intake ratios in rats and rabbits are generally several-fold higher (Buddington and Diamond 1990, Toloza and Diamond 1992), suggesting that growing chickens may function nearer their assimilation limit. Although such a match is a necessary condition for an assimilation bottleneck, it is not sufficient to demonstrate that one exists. A close match between physiological performance and a physiological need may indicate that a performance ceiling has been reached or, alternatively, it may simply indicate that regulatory mechanisms are acting effectively to reduce redundancy in the performance measure.

However, other lines of evidence suggest that heavy-bodied domestic chickens may be unusual compared to other vertebrates in normally operating very close to their assimilation capacity. While heavy-bodied chicken strains have been selected for rapid and efficient growth, light-bodied chickens (e.g. leghorns) have been selected for slower growth and efficient laying. Leghorn chickens can be overfed by up to 43%, and sustained force-feeding results in a large increase in intestine mass (56%) and augmented growth rates; heavy-bodied chicks (White Rock) can only be overfed by 11%, resulting in slight intestine growth (20%) and depressed (!) body growth (Nir et al. 1978). Similarly, light-bodied chicks respond to exposure to low ambient temperatures by increasing food intake and maintaining growth rates (Kleiber and Dougherty 1934); chicks of heavy-bodied strains keep food intake constant and grow more slowly under cold stress (Osbaldiston 1966). These independent lines of evidence suggest that selection for rapid and efficient production in heavy-bodied strains has selected for *ad libitum* feeding rates that approach the species' assimilation potential. This interpretation agrees with the close correspondence between uptake and intake we observed.

However, through most of its development, the capacity of the chick's intestine appears to be below the capacity that might have been achieved. This is evidenced by the intestine's ability suddenly (and reversibly) to increase its uptake capacity by several-fold. Furthermore, the sharp increases in nutrient uptake between weeks 1 and 2 and between weeks 5 and 6 did not coincide with periods of accelerated growth, but rather were associated with dips in the relative growth rate. However, the spikes appear to have set the stage for subsequent spurts in relative growth occurring in the third and seventh weeks of life (Fig. 1, right). This suggests that there may be an intrinsic lag in the intestine's ability to compensate adequately for sudden shifts in nutrient demand during development; alternatively, reorganization of the intestinal epithelium may itself be costly and occur at the temporary expense of body growth rates.

UNSOLVED PROBLEMS

Our results suggest the following areas for future research in the development of avian nutrient uptake:

1. Diet effects.—Domestic chickens have been selected to grow efficiently on diets of marginal quality (low protein). How might the general pattern of intestinal development that we observed be influenced by changes in diet already known to affect growth performance (i.e. diet restriction and shifts in relative proportions of carbohydrate, protein, and fat)?

2. Interspecific comparisons.—The intense artificial selection for rapid growth that makes the heavy-bodied chicken a useful model to test for assimilation bottlenecks also makes it difficult to generalize our conclusions. Do wild birds also operate near their uptake capacity? To answer this question, comparisons should be made in other avian species with a range of growth rates. Particularly interesting would be studies on other rapidly growing species, such as the precocial young of ducks and geese (Anatidae) whose natural growth rates are higher than their galliform counterparts, or altricial passerines, which exhibit the highest relative growth rates among vertebrates.

3. *Regulation.*—Ontogenetic changes in uptake in the chicken are often rapid and specific. How are these changes in nutrient transport physiologically mediated? Specifically, what are the signals (such as hormones or nutrient concentrations in the lumen) that cause sudden changes in one carrier independently of another, or in one region in preference to another? We believe that avian development promises to provide a useful model for future study of the regulation of phenotypic changes in carrier-mediated uptake.

4. *Why bottlenecks?*—If assimilation capacity limits growth, and if rapid growth has strong selective advantage, then why should such a bottleneck persist in the face of natural selection? Specifically, why should the growing chick not allocate even more tissue to its intestine or operate the intestine at maximal capacity throughout development? The costs associated with enhanced intestinal performance have yet to be defined.

ACKNOWLEDGMENTS

We wish to thank L. Cosko and K. Kumomoto for their valuable laboratory assistance in measuring nutrient uptake rates, and R. Jedas for generously providing software for the calculation of nutrient uptake rates. Funding for this project was provided by NIH grant (GM 14772) to JD and by a Public Health Service National Research Service Award (5 F32 DK07809) to BSO.

LITERATURE CITED

ALTMANN, G. G., AND M. ENESCO. 1967. Cell number as a measure of distribution and renewal of epithelial cells in the small intestine of growing and adult rats. Am. J. Anat. 121:319–336.

BOGNER, P. H., AND I. A. HAINES. 1964. Functional development of active sugar transport in the chick intestine. Am. J. Physiol. 207:37–41.

BUDDINGTON, R. K., AND J. M. DIAMOND. 1989. Ontogenetic development of intestinal nutrient transporters. Annu. Rev. Physiol. 51:601–619.

BUDDINGTON, R. K., AND J. M. DIAMOND. 1990. Ontogenetic development of nutrient transporters in rabbit intestine. Am. J. Physiol. 259:G544–G555.

BUDDINGTON, R. K., AND J. M. DIAMOND. 1992. Ontogenetic development of nutrient transporters in cat intestine. Am. J. Physiol. In press.

BUDDINGTON, R. K., J. W. CHEN, AND J. M. DIAMOND. 1987. Genetic and phenotypic adaptation of intestinal nutrient transport to diet in fish. J. Physiol. (Lond.) 393:261–281.

CARLOTTI, R. J. 1971. Manifestations of metabolic bursts during development of the cockerel. Ph.D. dissertation, West Virginia Univ., Morgantown.

CASE, T. J. 1978. On the evolution and adaptive significance of postnatal growth rates in the terrestrial vertebrates. Q. Rev. Biol. 53:243–282.

DIAMOND, J. M., AND K. A. HAMMOND. 1992. The matches, achieved by natural selection, between biological capacities and their natural loads. Experientia. In press.

FERRARIS, R. P., S. YASHARPOUR, K. K. LLOYD, R. MIRZAYAN, AND J. M. DIAMOND. 1990. Luminal glucose concentrations in the gut under normal conditions. Am. J. Physiol. 259:G822–G837.

HILL, R. W. 1972. Determination of oxygen consumption by use of the paramagnetic oxygen analyzer. J. Appl. Physiol. 33:261–263.

HOLDSWORTH, C. D., AND T. H. WILSON. 1967. Development of active sugar and amino acid transport in the yolk sac and intestine of the chicken. Am. J. Physiol. 212:233–240.

IMONDI, A. R., AND F. H. BIRD. 1966. The turnover of intestinal epithelium in the chick. Poult. Sci. 45:142–148.

JARVIS, L. G., G. MORGAN, M. W. SMITH, AND F. B. P. WOODING. 1977. Cell replacement and changing transport function in the neonatal pig colon. J. Physiol. (Lond.) 273:717–729.

KARASOV, W. H., AND J. M. DIAMOND. 1983. A simple method for intestinal solute uptake *in vitro*. J. Comp. Physiol. 152:105–116.

KARASOV, W. H., D. PHAN, J. M. DIAMOND, AND F. L. CARPENTER. 1986a. Food passage and intestinal nutrient absorption in hummingbirds. Auk 103: 453–464.

KARASOV, W. H., D. H. SOLBERG, S. CARTER, M. HUGHES, D. PHAN, F. ZOLLMAN, AND J. M. DIAMOND. 1986b. Uptake pathways for amino acids in mouse intestine. Am. J. Physiol. 251:G501–G508.

KATANBAF, M. N., E. A. DUNNINGTON, AND P. B. SIEGEL. 1988. Allomorphic relationships from hatching to 56 days in parental lines and F_1 crosses of chickens selected 27 generations for high or low body weight. Growth Dev. Aging 52:11–22.

KLEIBER, M., AND J. E. DOUGHERTY. 1934. The influence of environmental temperature on the utilization of food energy in baby chicks. J. Gen. Physiol. 17:701–726.

KONARZEWSKI, M., J. KOZLOWSKI, AND M. ZIOLKO. 1989. Optimal allocation of energy to growth of alimentary tract in birds. Funct. Ecol. 3:589–596.

KONARZEWSKI, M., C. LILJA, J. KOZLOWSKI, AND B. LEWONCZUK. 1990. On the optimal growth of the alimentary tract in avian postembryonic development. J. Zool. (Lond.) 222:89–101.

LACK, D. 1968. Ecological adaptations for breeding in birds. Metheun, London.

LERNER, J. 1984. Cell membrane amino acid transport processes in the domestic fowl (*Gallus domesticus*). Comp. Biochem. Physiol. 78A:205–215.

LERNER, J., AND D. L. STEINKE. 1977. Intestinal absorption of glutamic acid in the chicken. Comp. Biochem. Physiol. 57A:11-16.

LERNER, J., P. H. BURRILL, P. A. SATTLEMEYER, AND C. F. JANICKI. 1976. Developmental patterns of intestinal transport mechanisms in the chick. Comp. Biochem. Physiol. 54A:109-111.

LILJA, C. 1982. Postnatal growth and organ development in the Quail (*Coturnix coturnix japonica*). Growth 46:88-99.

LILJA, C. 1983. A comparative study of postnatal growth and organ development in some species of birds. Growth 47:317-399.

LILJA, C., I. SPERBER, AND H. L. MARKS. 1985. Postnatal growth and organ development in Japanese Quail selected for high growth rate. Growth 49: 51-62.

LUCAS, A. M., AND P. R. STETTENHEIM. 1972. Avian anatomy. Integument, Parts I and II. U.S. Department of Agriculture Handbook, No. 362.

MARCSTROM, V. 1966. Mallard ducklings (*Anas platyrhynchos*) during the first days after hatching: A physiological study with ecological considerations and a comparison with Capercaillie chicks (*Tetrao urogallus*). Viltrevy 4:343-370.

MARTIN, T. E. 1987. Food as a limit on breeding birds: A life history perspective. Annu. Rev. Ecol. Syst. 18:453-487.

MISSON, B. H. 1982. The thermoregulatory responses of fed and starved 1-week-old chickens (*Gallus domesticus*). J. Therm. Biol. 7:189-192.

MCCLELLAND, J. 1979. Digestive system. Pages 69-182 *in* Form and function in birds, vol. I (A. S. King and J. McClelland, Eds.). Academic Press, London.

NATIONAL RESEARCH COUNCIL. 1984. Nutrient requirements of poultry, 8th ed. National Academy Press, Washington, D.C.

NIR, I., AND Z. NITSAN. 1979. Metabolic and anatomical adaptations of light-bodied chicks to intermittent feeding. Br. Poult. Sci. 20:61-71.

NIR, I., Z. NITSAN, Y. DROR, AND N. SHAPIRA. 1978. Influence of overfeeding on growth, obesity, and intestinal tract in young chicks on light and heavy breeds. Br. J. Nutr. 39:27-35.

OBALDISTON, G. W. 1966. The response of the immature chicken to ambient temperature. Pages 228-234 *in* Physiology and biochemistry of the domestic fowl (C. Horton-Smith and E. C. Amoroso, Eds.). Oliver and Boyd, Edinburgh.

OBST, B. S., AND J. M. DIAMOND. 1989. Interspecific variation in sugar and amino acid transport by the avian cecum. J. Exp. Zool. Suppl. 3:117-126.

PEACH, H. C., AND V. G. THOMAS. 1986. Nutrient composition of yolk in relation to early growth of Canada Geese. Physiol. Zool. 59:344-356.

PLANAS, J. M., M. MORETO, E. GAZA, AND J. BOLUFER. 1982. Changes in intestinal galactose and leucine transport during development of the chick: Effect of low external calcium. Poult. Sci. 61:1094-1098.

PLANAS, J. M., M. C. VILLA, R. FERRER, AND M. MORETO. 1986. Hexose transport by chicken cecum during development. Pflügers Arch. Eur. J. Physiol. 407: 216-220.

RAHEJA, K. L., J. TEPPERMAN, AND H. M. TEPPERMAN. 1977. The effect of age on intestinal glucose transport in the chick (*Gallus domesticus*). Comp. Biochem. Physiol. 58A:245-248.

RICKLEFS, R. E. 1968. Patterns of growth in birds. Ibis 110:419-451.

RICKLEFS, R. E. 1969. An analysis of nestling mortality in birds. Smithson. Contrib. Zool. 9:1-48.

RICKLEFS, R. E. 1973. Patterns of growth in birds. II. Growth rate and mode of development. Ibis 115: 177-201.

RICKLEFS, R. E. 1976. Growth rates of birds in the humid New World tropics. Ibis:179-207.

RICKLEFS, R. E. 1979. Adaptation, constraint, and compromise in avian postnatal development. Biol. Rev. Camb. Philos. Soc. 54:269-290.

RICKLEFS, R. E. 1983. Avian postnatal development. Pages 1-83 *in* Avian biology, vol. VII (D. S. Farner and J. R. King, Eds.). Academic Press, New York.

ROMANOFF, A. L. 1944. Avian spare yolk and its assimilation. Auk 61:235-241.

SHEHATA, A. T., J. LERNER, AND D. S. MILLER. 1981. Development of brush-border membrane hexose transport system in chick jejunum. Am. J. Physiol. 240:G102-G108.

SMITH, M. W., AND L. G. JARVIS. 1978. Growth and cell replacement in the newborn pig intestine. Proc. R. Soc. Lond. Ser. B Biol. Sci. 203:69-89.

SOKAL, R., AND F. J. ROHLF. 1981. Biometry, 2nd ed. W. H. Freeman, San Francisco.

TOLOZA, E. M., AND J. M. DIAMOND. 1990. Ontogenetic development of nutrient transporters in bullfrog intestine. Am. J. Physiol. 258:G760-G769.

TOLOZA, E. M., AND J. M. DIAMOND. 1992. Ontogenetic development of nutrient transporters in rat intestine. Am. J. Physiol. In press.