Low intrinsic running capacity is associated with reduced skeletal muscle substrate oxidation and lower mitochondrial content in white skeletal muscle.

Donato A. Rivas¹, Sarah J. Lessard¹, Misato Saito², Anna M. Friedhuber³, Lauren G. Koch⁴, Steven L. Britton⁴, Ben B. Yaspelkis III², John A. Hawley¹

¹ Health Innovations Research Institute, School of Medical Sciences, RMIT University, Bundoora, Australia; ² Department of Kinesiology, California State University, Northridge, CA, USA; ³ Department of Pathology, University of Melbourne, Australia; ⁴ Department of Physical Medicine and Rehabilitation, University of Michigan, Ann Arbor, MI, USA

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Address for correspondence:

John A. Hawley, Ph.D.
Health Innovations Research Institute
School of Medical Sciences
RMIT University
PO Box 71
Bundoora
VIC 3083
AUSTRALIA
Phone: +61-3-9925 7353
Fax: +61-3-9467 8181
e-mail: john.hawley@rmit.edu.au
Abstract

Background
Chronic metabolic diseases develop from the complex interaction of environmental and genetic factors, although the extent to which each contributes to these disorders is unknown. Here we test the hypothesis that artificial selection for low intrinsic aerobic running capacity is associated with reduced skeletal muscle metabolism and impaired metabolic health.

Methodology/Principal Findings
Rat models for low- (LCR) and high- (HCR) intrinsic running capacity were derived from genetically heterogeneous N:NIH stock for 20 generations. Artificial selection produced a 530% difference in running capacity between LCR/HCR, which was associated with significant functional differences in glucose and lipid handling by skeletal muscle, as assessed by hind limb perfusion. LCR had reduced rates of skeletal muscle glucose uptake (~30%; P=0.04), glucose oxidation (~50%; P=0.04) and lipid oxidation (~40%; P=0.02). Artificial selection for low aerobic capacity was also linked with reduced molecular signaling, decreased muscle glycogen and triglyceride storage and a lower mitochondrial content in skeletal muscle, with the most profound changes to these parameters evident in white rather than red muscle.

Conclusions/Significance
We show that a low intrinsic aerobic running capacity confers reduced insulin sensitivity in skeletal muscle and is associated with impaired markers of metabolic health compared to high intrinsic running capacity. Furthermore, selection for high running capacity, in the absence of exercise training, endows
increased skeletal muscle insulin sensitivity and oxidative capacity in specifically white muscle rather than red muscle. This data provides evidence that differences in white muscle may have a role in the divergent aerobic capacity observed in this generation of LCR/HCR.

**Keywords:** genetics; insulin signaling; mitochondria; oxidative capacity; skeletal muscle
Introduction

During the past 50 years, the prevalence of a cluster of interrelated chronic metabolic disease states including coronary heart disease, insulin resistance, type 2 diabetes mellitus (T2DM) and obesity has reached epidemic proportions (2). The etiological basis of these disorders is polygenic and highly dependent on the environment (i.e. existing genes interact with environmental factors to result in phenotypic expression of these diseases). One environmental factor to have changed dramatically in this time and strongly associated with a plethora of chronic metabolic disorders is the decline in physical activity (8). Indeed, the increased prevalence of cardiovascular disease, insulin resistance, type 2 diabetes and obesity and their strong association with inactivity has produced an “exercise deficient phenotype” in which individuals with a particular combination of disease-susceptible genes (i.e. risk factors) interact with undefined environmental conditions (e.g., level of physical activity) and cross a threshold of biological significance that results in overt clinical conditions. Evidence in support of this premise comes from studies in which multiple genes involved in aerobic metabolism are down regulated in several metabolic states and may be linked to the pathogenesis of these disorders (31, 38).

Skeletal muscle plays a vital role in locomotion and makes an important contribution to whole-body energy metabolism, disposing of up to ~80% of a postprandial glucose load (7, 56). Whole-body metabolic health is associated with the ability of skeletal muscle to transition between the uptake and oxidation of carbohydrate- and lipid-based fuels in response to their availability and the prevailing hormonal milieu (20). In several lifestyle-related diseases such as
obesity and type 2 diabetes, there is a loss of skeletal muscle plasticity such that rates of substrate oxidation do not increase effectively in response to fuel availability (10, 20, 22). Impaired muscle oxidative capacity coupled with a decreased ability of muscle to oxidize lipids are strong predictors of insulin resistance (3, 42). In support of this premise, several groups have reported that skeletal muscle mitochondrial content, mitochondrial function and/or oxidative capacity are reduced in individuals that are insulin resistant or have type 2 diabetes (T2DM) (15, 18, 21, 42). Taken collectively, the results from these studies suggest that the lower oxidative capacity observed in individuals with obesity, T2DM, or both, may play a significant functional role in the development of insulin resistance. However, it is not known to what extent low oxidative capacity in muscle and the concomitant insulin resistance are a result of genetically predetermined or environmental factors.

In order to identify the genetic contribution to oxidative capacity, we have developed a novel rat model through two-way artificial selection for either low (LCR) or high (HCR) aerobic treadmill running capacity (24). After 11 generations of selective breeding, there was a 374% difference in running capacity between phenotypes that was associated with an increase in risk factors for cardiovascular disease, such as hyperinsulinemia, hyperlipidemia and increased adiposity in LCR compared to HCR (36, 52). HCR rats were also resistant to the development of high-fat diet-induced obesity and insulin resistance (35, 36).

The aim of the present investigation was to test the hypothesis that low intrinsic aerobic running capacity is associated with impaired skeletal muscle oxidative capacity and blunted insulin sensitivity. This was accomplished by
mimicking a “fasted” and “fed” environment in the skeletal muscle of the LCR and HCR and determining rates of insulin-stimulated glucose uptake and oxidation and basal lipid uptake and oxidation using hind limb perfusion techniques. We have attempted to elucidate potential mechanisms that impart the impaired metabolic health associated with a low intrinsic running capacity by measuring substrate storage, insulin signal transduction and mitochondrial activity and density in both oxidative and glycolytic muscle, from these divergent phenotypes.

**Methods**

*Experimental Animals*

Forty-eight female LCR/HCR rats from generation 20 (15-16 wk old) and 22 (20 wk old) (G20 and G22) were used. Rats models for high and low aerobic capacity were derived from genetically heterogeneous N:NIH stock rats by artificial selection for low and high aerobic running capacity (52). Animals were phenotyped for intrinsic running capacity at 11 wk of age using an incremental running test with the treadmill constantly at an uphill incline of 15° (52). Rats were housed two per cage in a temperature-controlled animal room (21ºC) maintained on a 12-hour light-dark cycle. Animals were provided with standard chow diets and water *ad libitum*. All animal experimentation procedures were carried out with the approval of animal ethics committees from California State University, Northridge; the University of Michigan; and RMIT University (AEC 0805).

*Blood Measures*

Fasting blood values were taken after a 5 h fast and glucose concentrations were determined with the MediSense2 Blood Glucose Testing system
(MediSense Australia; Melbourne, Australia). Fasting serum non-esterified fatty acid measures were obtained using an enzymatic colorimetric method (NEFA C; Wako Pure Chemicals, Osaka, Japan).

**Glucose Tolerance Test**

Animals from G20 were fasted for 5 h before receiving an intraperitoneal injection of D-glucose (1 g/kg body mass). Blood glucose concentrations were measured at 0, 15, 30, 45, 60, 90 and 120 min following the glucose dose. The area under the blood glucose curve (AUC; mM*min) was calculated for each animal.

**Hind limb Perfusions**

Animals from G20 were fasted for 5-7 h before undergoing hind limb perfusion for the measurement of insulin-stimulated D-[\textsuperscript{14}C(U)]-glucose uptake/oxidation (n=8/group) or [\textsuperscript{14}C]-palmitate uptake/oxidation (n=8/group). Rats were anesthetized and surgically prepared for hind limb perfusion as previously described (26, 54). Just prior to cannulation, portions of the red (RG) and white (WG) gastrocnemius were excised from the non-perfused left leg, freeze clamped in liquid N\textsubscript{2}, and stored at -80\textdegree C until later analysis. In the non-perfused RG and WG, citrate synthase activity, intramuscular triacylglycerol, glycogen content and carnitine palmitoyltransferase I (CPTI) protein content were assessed. The basic perfusate medium consisted of 30% washed time-expired human erythrocytes (Ogden Medical Center, Ogden, UT, USA), 4% dialysed bovine serum albumin (Equitech-Bio Inc., Kerrville, TX USA) and Krebs–Heinseleit Buffer (KHB) [pH 7.4]. The perfusate was continuously gassed with a mixture of 95% O\textsubscript{2}/5% CO\textsubscript{2} and warmed to 37\textdegree C.

**D-[\textsuperscript{14}C(U)]-glucose uptake/oxidation rates**
Immediately after cannulation, the rats were sacrificed via an intracardiac injection of pentobarbital while the hind limbs were washed out with 20 ml of heparinised (10 U/ml) KHB. The catheters were then placed in line with a non-recirculating perfusion system, and the hind limb was allowed to stabilize during a 5 min washout period. The perfusate flow rate was set at 5 mL/min during the 5 min stabilization period and subsequent perfusion. Perfusions were performed in the presence of insulin (1 mU/mL) for all experimental groups. Following the stabilization period, the perfusate was changed to one containing 8 mM glucose (0.25 μCi/ml D-[\textsuperscript{14}C(U)] glucose, PerkinElmer Life Sciences, MA, USA). At the completion of the 30 min perfusion, portions of the RG and WG were removed, blotted on gauze dampened with cold KHB, clamp frozen in liquid N\textsubscript{2} and stored at -80°C until later analysis. For determination of glucose uptake, perfusate samples were taken from the arterial perfusate and well-mixed venous effluent, deproteinized in 10% TCA, centrifuged and quantified using liquid scintillation counting. Muscle glucose uptake was calculated from the arteriovenous difference, the perfusate flow rate and the weight of the muscle perfused.

Arterial and venous samples for the analysis of \textsuperscript{14}CO\textsubscript{2} were taken immediately at the end of a 5 min equilibration period and at the end of the perfusion (30 min). The liberation and collection of \textsuperscript{14}CO\textsubscript{2} from perfusate samples was performed by injecting 2 ml of anaerobically collected perfusate into a sealed flask containing an equal volume of 1 M acetic acid. The released \textsuperscript{14}CO\textsubscript{2} was trapped by an insert containing a strip of filter paper saturated with 500 μl benzethonium hydroxide and quantified using liquid scintillation counting. The rate of glucose oxidation was determined from the arteriovenous
difference in $^{14}$CO$_2$ and flow rate. Lactate was determined as previously described (54). Lactate accumulation was calculated from the arteriovenous difference, perfusate flow rate and the weight of the muscle perfused.

[$^{14}$C] palmitate uptake/oxidation rates

Skeletal muscle fatty acid metabolism was assessed as previously described (26). In brief, immediately after cannulation, the rats were sacrificed via an intracardiac injection of pentobarbital while the hind limbs were washed out with 20 ml of heparinized (10 U/ml) KHB. The catheters were then placed in line with a recirculating perfusion system, and the hindlimb was allowed to equilibrate during a 20 min period. The perfusate flow rate was set at 7.5 ml/min during the 50 min perfusion (20 min equilibration / 30 min perfusion) with perfusate containing 4% fatty acid-free BSA (Equitech-Bio Inc.), 500 $\mu$mol/l albumin-bound palmitate (Sigma Aldrich; MO, USA), and 5 $\mu$Ci albumin-bound $[1-^{14}$C] palmitate (PerkinElmer; MA, USA). For determination of palmitate uptake, perfusate samples were anaerobically taken from the arterial perfusate and venous lines, centrifuged and quantified using liquid scintillation counting. Muscle palmitate uptake was calculated from the arteriovenous difference, the perfusate flow rate and the weight of the muscle perfused.

Arterial and venous samples for the analysis of $^{14}$CO$_2$ were taken immediately after a 20 min equilibration period and at 50 min. The liberation and collection of $^{14}$CO$_2$ from perfusate samples was performed by injecting 2 ml anaerobically collected perfusate into a sealed flask containing an equal volume of 1M acetic acid. The released $^{14}$CO$_2$ was trapped by an insert containing a strip of filter paper saturated with 500 $\mu$l benzethonium hydroxide and quantified using liquid scintillation counting.
Analysis of intramuscular substrate storage and citrate synthase activity

Portions of RG and WG muscle were freeze dried, powdered and analysed for the content of glycogen, glycerol, and maximal citrate synthase activity as previously described (26, 55). Briefly, freeze-dried muscle was powdered and cleaned of all visible connective tissue and blood under magnification. Portions (4-5 mg) of freeze dried, powdered RG (8-10/group) and WG (8-10/group) were used to fluorometrically determine skeletal muscle triacylglycerol (total glycerol) content, following Folch lipid extraction and saponification. Portions (3-4 mg) of freeze dried, powdered RG (8-10/group) and WG (8-10/group) were used to enzymatically analyse glycogen content by fluorometric detection after extraction with 2M HCl and neutralization with 0.67M NaOH. Portions (3-4 mg) of freeze dried, powdered RG (8-10/group) and WG (8-10/group) were homogenized in 100 mM potassium phosphate buffer (pH 7.3, 1:400 dilution), and citrate synthase was assayed spectrophotometrically at 25°C by the reduction of DTNB.

Western blotting analysis

Portions of muscle were cut from the RG and WG, weighed frozen and homogenized in an ice-cold homogenization buffer (1:8 wt/vol) containing 50 mM Tris-HCl (pH 7.5), 5 mM Na-pyrophosphate, 50 mM NaF, 1 mM EDTA, 1 mM EGTA, 10% glycerol (v/v), 1% Triton-X, 1 mM DTT, 1 mM benzamidine, 1 mM PMSF, 10 μg/ml trypsin inhibitor, and 2 μg/mL aprotinin. Following centrifugation (21,000 x g, 4° C) for 15 min the supernatant was collected and assayed for protein content. RG muscle lysates (60 μg) were solubilized in Laemmli buffer, separated by SDS-PAGE, and transferred to PVDF membranes. The membranes were then blocked (5% NFDM), and incubated
overnight at 4°C with primary antibodies specific for either phospho-Akt Ser473, phospho-Akt Thr308, Akt1, Akt2, phospho-Akt substrate (1:1000; Cell Signaling, MA, USA), phospho-IRS1 Tyr632, CPTI (1:200-500; Santa Cruz Biotechnology, CA, USA), or AS160 (TBCD14) which was produced as previously described (25) using a region of human AS160 from amino acids 621-766 fused with GST (1:1000, a gift from Prof. David James, Garvan Institute, Sydney, Australia). The immunoreactive proteins were detected with enhanced chemiluminescence (Amersham Biosciences) and quantified by densitometry.

*Transmission electron microscopy (EM) for determination of mitochondrial content*

Soleus (SOL) [4/group] and extensor digitorum longus (EDL) [4/group] skeletal muscle from a separate cohort of animals (G22) were excised, cut into small longitudinal strips (1x1x2 mm) fixed in 2.5% glutaraldehyde in 0.1M cacodylate buffer (pH 7.4) and postfixed in 2% osmium tetroxide solution. After dehydrating in graded acetone, tissue was embedded in araldite/epon resin. Thick sections (0.5 μm) were cut using Ultracut S ultramicrotome (Leica, North Ryde, New South Wales, Australia) and then stained with 1% methylene blue. Thin sections (90 nm) were cut using the same microtome mounted on copper/palladium 200 mesh grids and then stained with 3% aqueous solution of uranyl acetate and lead citrate (Reynold’s stain). Randomly sampled transverse sections of muscle fibers were obtained followed by micrographs acquired with an electron microscope at 60V (Siemens Elmiskop 102 electron microscope) on a final magnification of 10,000x. In order to obtain a valid representation of the whole muscle, two micrographs (1 from the subsarcolemmal region and 1 from
the adjacent interfibrillar region) in three separate muscle fibers for each muscle (SOL, EDL) in each group were acquired for a total of 6 micrographs/muscle/animal. Mitochondrial content was determined using the point-counting stereological analysis methodology with Image J software (Image J 1.41, National Institutes of Health, USA). Each micrograph was counted and then recounted in a double-blind fashion.

**Statistical Analysis**

Differences between LCR and HCR were identified using a two-tailed t-test with GraphPad Prism version 4.04 for Windows (GraphPad Software, CA, USA, www.graphpad.com). Results are expressed as mean ± SD and statistical significance was accepted at P<0.05.

**Results**

*Markers of metabolic health are divergent in HCR and LCR.*

Intrinsic treadmill running capacity was 530% greater in HCR compared to LCR (Generation 20, P=0.0001, Table 1). HCR had lower body mass and fasting serum glucose levels (P<0.0002 vs. LCR; Table 1), but had no difference in the levels of circulating free-fatty acids (FFA) compared to LCR. HCR were more glucose tolerant than LCR, as measured from the AUC after an intraperitoneal glucose tolerance test (IPGTT) following a 5 h fast (P=0.01; Table 1).

*HCR have superior substrate handling in skeletal muscle.*

To determine if intrinsic differences in aerobic capacity were associated with alterations in glucose and lipid handling, “fasted” and “fed” states were mimicked during hind-limb perfusions. HCR had superior insulin-stimulated glucose metabolism as demonstrated by a 30% higher rate of glucose uptake
(P=0.04 vs. LCR; Figure 1A) and a 50% higher rate of glucose oxidation (P=0.04 vs. LCR; Figure 1B). HCR also had superior lipid metabolism, with a higher rate of palmitate oxidation (P=0.02 vs. LCR; Figure 1D), and a trend for a higher palmitate uptake (P=0.10 vs. LCR; Figure 1C). Lactate accumulation during the 30 min perfusion was similar for both phenotypes (4.464 ± 1.2μmol/g/h vs. 4.052 ± 0.7μmol/g/h).

Substrate storage in skeletal muscle was greater in HCR.

The intramuscular storage of glycogen and lipid was determined in basal (unstimulated) tissue. There were no differences in glycogen storage in the RG between LCR and HCR, but there was a 21% increase in glycogen content in the WG of HCR (P=0.042 vs. LCR; Figure 2A). IMTG concentration was 18% higher in the RG of HCR (P=0.04 vs. LCR; Figure 2B).

Enhancement of insulin signaling in WG muscle of HCR.

In order to determine insulin-stimulated cell signalling responses in white and red muscle, the phosphorylation of proximal and distal components of the insulin signalling cascade were assessed. There were no differences between phenotypes in the phosphorylation of IRS1 on tyrosine (Y)632 in the RG. However, there was an increase in IRS1 phosphorylation in the WG of HCR (P=0.002 vs. LCR; Figure 3A). The phosphorylation of Akt on threonine (T)308 and serine (S)473 was also greater in the white muscle from HCR compared to LCR with a 24% and 26% increase in the WG (P=0.016, P=0.03 vs. LCR; Figure 3B, 3C), but not RG. The phosphorylation of AS160, a downstream substrate of Akt, was also greater in WG (21% increase, P=0.03 vs. LCR; Figure 3D) and tended to be higher in the RG. (P=0.12 vs. LCR; Figure 3D). There was a 46% increase in the expression of the Akt1 isoform in the RG of
HCR (P=0.02 vs. LCR; Figure 3E) and a 95% increase in the WG (P=0.03 vs. LCR; Figure 3E). In accordance with these observations, the expression of Akt2 was also increased in the RG (35%, P=0.026 vs. LCR; Figure 3E) and in the WG (47%, P=0.008 vs. LCR; Figure 3E) of HCR. There was a decrease in the expression of AS160 in the RG (P=0.04 vs. LCR; Figure 3E), but no change in the WG of the HCR. There were no differences in RG and WG when phosphorylated Akt/AS160 was normalized to total Akt/AS160 for either group.

Enhanced mitochondrial oxidative capacity in white but not red muscle of HCR.

In order to determine whether there were differences in markers of mitochondrial capacity and density between phenotypes, citrate synthase activity, the expression of mitochondrial proteins and mitochondrial content as quantified by EM were determined. There were no differences in the maximal activity of citrate synthase in RG of HCR and LCR, but citrate synthase activity was increased in WG of HCR (51%, P=0.0004 vs. LCR; Figure 4A). There were no differences in the expression of CPTI in the RG for either phenotype (Figure 4B). However, the expression CPTI tended to be higher (P=0.098 vs. LCR; Figure 4B). Representative EM images of Sol and EDL of LCR/HCR are shown in Figure 5A. There was no significant difference in mitochondrial content in the SOL (a red, oxidative muscle) between groups (Figure 5B). However, there was a ~70% higher mitochondrial content in the EDL (a white, glycolytic muscle) of the HCR (P=0.0002 vs. LCR; Figure 5B).
Discussion

Divergent selection for low- and high intrinsic aerobic capacity has generated a powerful model system for dissection of the role of aerobic endurance capacity and its correlated traits. Importantly, simultaneous breeding of the LCR and HCR at each generation also allows for each line to serve as a control for unknown environmental changes. The LCR/HCR model of divergent intrinsic aerobic capacity was developed to specifically study some of the potential mechanisms associated with the metabolic syndrome and cardiovascular disease in a system in which there is a degree of control over both environmental and genetic variables. Hence the results from the present investigation provide novel insight into the fundamental molecular events underlying increased risk for metabolic disease.

The inability of skeletal muscle to adjust fuel oxidation to fuel availability is strongly associated with the development and progression of several chronic disease states such as obesity, type 2 diabetes and cardiovascular disease (10, 20). Utilizing hind-limb perfusion techniques, we demonstrate that genetic selection for low intrinsic running capacity was associated with decreased rates of skeletal muscle glucose uptake (~30%; P=0.04; Fig 1A), glucose oxidation (~50%; P=0.04; Fig 1B) and lipid oxidation (~40%; P=0.02; Fig 1D), as well an increase in whole-body metabolic markers of disease risk (Table 1). In contrast, high intrinsic aerobic capacity imparts an increased capacity to utilize introduced substrates (Fig 1) and is linked with improvements in whole-body markers of metabolic health (Table 1). Artificial selection for high running capacity was further linked with superior molecular signalling, substrate storage and mitochondrial content in skeletal muscle.
Increased storage of lipids in insulin sensitive tissue, such as skeletal muscle, has been shown to be a primary contributor to decreased aerobic capacity, insulin resistance and impaired glycogen synthesis (5, 12, 21, 26, 39). These defects are believed to precede type 2 diabetes and are thought to have a primary contribution to this disease state. In the present investigation we report that the lower insulin sensitivity in LCR was associated with reduced glycogen storage in muscle (Fig 2A) but not increased lipid storage (Fig 2B). We also report an increase in TAG in muscle from HCR animals (Fig 2B). While this observation appears paradoxical, TAG levels are increased in the muscle of endurance trained athletes: these individuals also have high muscle oxidative capacity and are highly insulin sensitive, a finding that has been termed an “athlete's paradox” (11). We have also previously observed that oxidative capacity in skeletal muscle is a better predictor of insulin sensitivity than intramyocellular lipid content in insulin resistant individuals (3). Thus, the skeletal muscle of HCR demonstrates similar properties to muscle of endurance trained athletes, albeit in the total absence of exercise training. These results suggest there may be a strong genetic contribution to the enhanced aerobic capacity of endurance athletes.

Impairments to insulin signal transduction are strongly associated with impaired carbohydrate and lipid metabolism in skeletal muscle (6, 10, 13). Insulin resistance in skeletal muscle is associated with impairments in the activation, phosphorylation, and expression of major components of the insulin signaling cascade (13, 19, 40). However, the exact mechanisms associated with impaired insulin-stimulated carbohydrate metabolism and signal transduction in low aerobic capacity has yet to be determined. Here we show
for the first time that low aerobic capacity is associated with the reduced phosphorylation to components of the insulin signal cascade. In particular, we observed a fiber-type specific impairment in the phosphorylation of IRS1 on Y632, being lower in the WG of the LCR compared to HCR (Fig 3A). This effect was also seen downstream of IRS1 with the reduced phosphorylation of Akt on its T308 (Fig 3B) and S473 (Fig 3C) sites and reduced phosphorylation of its substrate AS160 (Fig 3D) in WG. Although the decrease in Akt phosphorylation was likely a result in the reduced protein content in LCR rather than decreases in kinase activity, these differences in total protein content changed the potential for the total amount of enzyme activity in the cell.

In the present investigation, we have analyzed both red (soleus and red gastrocnemius) and white (extensor digitorum longus and white gastrocnemius) muscle to determine whether the metabolic phenotype of LCR/HCR is fiber-type specific. Differences in the physiological properties and function of red and white skeletal muscle have been well characterized (14, 29, 46, 50). Red muscle has a higher oxidative capacity, partly because of higher concentrations of mitochondria, and is more insulin sensitive than white muscle (17, 37). In response to endurance exercise training, several kinases have been found to be differentially activated in red and white skeletal muscle (28, 34) and insulin signalling has been found to be impaired in both fiber types in the skeletal muscle of diabetic animals (45).

A novel finding from the present study was that the most significant changes in mitochondrial content occurred in white muscle rather than in red muscle between the divergent phenotypes (Fig 5B). We found a significantly lower mitochondrial content in the EDL muscle from LCR (P<0.001 vs. HCR;
Fig 5B), but no significant difference in the soleus (Fig 5B). These findings paralleled the decrease in CS activity (Fig 4A) observed in the WG of LCR. Interestingly, other groups, using genetically manipulated mouse models (transgenic or knockout for PGC1α, PPARδ or Calsarcin-2), have also observed a fiber-type shift with white glycolytic muscle taking on the functional properties of red fibers (e.g., increased myoglobin, SDH activity, fatigue resistant, etc.). Such fibre-type changes were positively correlated with an increased endurance capacity (9, 27, 51). Hood et al. (16) have previously noted that the coordination of metabolic plasticity (i.e., mitochondrial content) can, in large part, be attributed to change in the properties of white glycolytic as opposed to red oxidative muscle. Interestingly, it was recently observed in generation 18 of LCR/HCR that there is a tendency for a more oxidative phenotype in the EDL of HCR (23) which is consistent with our present finding of an increased mitochondrial content in this muscle. The similarity in mitochondrial content in the soleus muscle between phenotypes observed in the present study may, in part, be due to its constant loading in caged animals (53).

Reduced mitochondrial content in the skeletal muscle of LCR was associated with increased body weight, decreased fatty acid oxidation and reduced insulin sensitivity in these animals. These results are in agreement with the fact that in obesity and insulin resistance there is impairment to mitochondrial oxidative capacity in skeletal muscle (30, 44, 48). Furthermore, insulin resistant subjects with a family history of diabetes have low mitochondrial oxidative capacity that is correlated with decreased mitochondrial density in skeletal muscle as measured by electron microscopy (1, 32). We
have previously reported a reduction in PPARγ coactivator 1α (PGC1α) protein abundance and in the concentration of the oxidative enzyme, cytochrome c oxidase subunit 1 (COX1), in skeletal muscle from LCR (36). Reductions in the expression or activation of mitochondrial enzymes or proteins, like PGC1α, CPTI or CS, are related to a decline in the oxidative capacity of skeletal muscle (4, 33, 43).

Our investigation demonstrates that selection for the trait of low intrinsic aerobic capacity alters the ability of skeletal muscle to oxidize substrate in response to their availability. The LCR/HCR models used in the present investigation provide novel evidence of a genetic contribution to muscle oxidative capacity. This is in agreement with previous studies that have reported data suggesting that reduced oxidative capacity may represent an early stage in the progression of metabolic disease states (ie. insulin resistance) in some humans, and that low oxidative capacity may be an inherited defect in such people (32, 48, 49). Indeed rates of lipid oxidation at rest have been observed to be persistently low even in formerly obese people who subsequently lose weight (41, 47).

**Perspectives and Significance**

In conclusion, selection for low intrinsic aerobic running capacity was associated with reduced insulin signalling, decreased skeletal muscle oxidative capacity and lowered mitochondrial content. The impaired fuel-handling capacity in animals bred for inferior running capacity can, in part, be explained by a fiber-type specific decrease in insulin-stimulated phosphorylation of the insulin receptor substrate 1 (IRS1), Akt and the Akt substrate of 160 kDa (AS160) along with a substantial decrease in mitochondrial content. Using
skeletal muscle-specific measures of substrate metabolism, our results provide the first evidence that low intrinsic running capacity is associated with impaired oxidative capacity and substrate handling and decreased insulin signalling and mitochondrial content. Furthermore, selection for high running capacity, in the absence of exercise training, endows increased skeletal muscle insulin sensitivity and oxidative capacity, the latter of which is mainly confined to white rather than red muscle. This data provides some evidence that differences in the white muscle may have a role in the divergence in aerobic capacity observed in this generation of LCR/HCR.
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Current Address DAR: Nutrition, Exercise Physiology and Sarcopenia Laboratory; Jean Mayer USDA Human Nutrition Research Center on Aging; Tufts University; 711 Washington Street, Boston, MA, United States 02111.

Current address for SJL: Joslin Diabetes Center, Harvard Medical School, 1 Joslin Place, Boston, Massachusetts 02215.
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Table 1. Whole-body characteristics of HCR and LCR models (n=10/group).

* Significant differences between groups (P<0.05)

Figure 1. Skeletal muscle glucose and lipid metabolism was assessed in generation 20 LCR and HCR after a 5 hour fast. Average \[^{14}\text{C}\] glucose uptake (A) and \[^{14}\text{C}\]CO\(_2\) production due to \[^{14}\text{C}\] glucose oxidation (B) were calculated using differences in arterial and venous perfusate concentrations during a 30 min hind limb perfusion with 8 mM glucose and 1 mU/mL insulin. Average \[^{14}\text{C}\] palmitate uptake (C) and \[^{14}\text{C}\]CO\(_2\) production due to \[^{14}\text{C}\] palmitate oxidation (D) were calculated using differences in arterial and venous perfusate concentrations during a 30 min hind limb perfusion with 1.8 mmol/l palmitate. Significant differences between groups (P<0.05) are indicated by the P value listed on the figure (n=7-8/group).

Figure 2. Intramuscular glucose and lipid storage. Muscle glycogen (A) and triacylglycerol (B) content were determined in red and white gastrocnemius on separate aliquots of freeze dried/powdered muscle and expressed per mg of dry weight [DW]. Significant differences between groups (P<0.05) are indicated by the P value listed on the figure (n=8-10/group).

Figure 3. Insulin-stimulated cell signalling in skeletal muscle. Phosphorylation of IRS1 on Tyr632 (A), Akt on Thr308 (B) and Ser473 (C), and PAS160 (D) and relative total protein levels of Akt1, Akt2 and AS160 (E) were determined by western blotting on insulin-stimulated perfused red and white gastrocnemius muscle. To confirm equal loading, \(\alpha\)-tubulin was run as a loading control. Significant differences between groups (P<0.05) are indicated by the P value listed on the figure or *(P<0.05) next to the representative blot (n=7-8/group).

Figure 4. Mitochondrial enzyme activity and mitochondrial protein content in skeletal muscle. Citrate synthase activity (A) as determined by enzymatic assay and the relative protein levels of CPTI (B) as quantified using western blot analysis and densitometry were analysed in red and white gastrocnemius muscle. To confirm equal loading, in western blots, \(\alpha\)-tubulin was run as a loading control. Significant differences between groups (P<0.05) are indicated by the P value listed on the figure (n=7-8/group).

Figure 5. Mitochondrial content as determined by electron microscopy in soleus (n=4/group) and extensor digitorum longus (n=4/group). Representative micrographs (A) of soleus (Sol) and extensor digitorum longus (EDL) skeletal muscle on transverse sections. Mitochondrial content was determined by point-count method in the subsarcolemmal (SS) and intermyofibrillar (IMF) regions of the Sol (B) and EDL (C). Significant differences between groups (P<0.05) are indicated by the P value listed on the figure (n=48 micrographs/group).
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<td>Initial Running Capacity (m)</td>
<td>282.4 ± 59.6</td>
<td>1776 ± 175.4*</td>
</tr>
<tr>
<td>Non-Esterified Fatty Acids (mM)</td>
<td>0.582 ± 0.28</td>
<td>0.384 ± 0.06</td>
</tr>
<tr>
<td>Fasting Glucose (mmol/l)</td>
<td>6.23± 0.51</td>
<td>5.18± 0.64 *</td>
</tr>
<tr>
<td>Body Weight (g)</td>
<td>227.7 ± 22.1</td>
<td>166.3 ± 22.6*</td>
</tr>
<tr>
<td>Glucose Tolerance Test (mM*min)</td>
<td>916.9 ± 50.1</td>
<td>828.7 ± 95.3 *</td>
</tr>
</tbody>
</table>

* P<0.05 vs. LCR
A  Glucose Uptake

B  Glucose Oxidation

C  Palmitate Uptake

D  Palmitate Oxidation

P = 0.04

P = 0.04

P = 0.10

P = 0.02
A

Intramuscular Glycogen

- **RG**
- **WG**

![Bar graph showing intramuscular glycogen levels for LCR and HCR, with RG and WG groups, and statistical significance P=0.04.]

B

Intramuscular Tryglycerides

- **RG**
- **WG**

![Bar graph showing intramuscular tryglycerides levels for LCR and HCR, with RG and WG groups, and statistical significance P<0.05.]
A

Citrate Synthase Activity

B

CPT I

α-tubulin

RG

WG

P<0.001

P<0.10

CPT I (Arbitrary Units)

CPT I (Arbitrary Units)