# Muscle mTORC1 Activation Causes Reduced Adiposity in Mice

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# Abstract

# Introduction

Obesity is a worldwide health problem, with comorbidities including diabetes, cardiovascular and liver disease [1]. Current modalities to prevent or reverse obesity are ineffective and short-lived often due to reductions in energy expenditure and increases in hunger after weight loss [2–4]. The genetic and dietary modifiers of energy expenditure are not well known, but recent reports have implicated low-carbohydrate (or high protein) diets in enhancing energy expenditure in both overfeeding and weight loss paradigms [5,6]. The mechanism(s) by which these diets have these effects have not yet been completely elucidated.

mTORC1 is a nutrient responsive protein kinase complex expressed in all known eukaryotic cells. This complex is activated by anabolic signals such as insulin, amino acids and energy abundance (see [7] for review). mTORC1 integrates these signals, and helps to co-ordinate such anabolic processes as lipogenesis [8–10], glycogenesis [11], cellular differentiation [12–14] while promoting insulin resistance [8,15]. These effects are often tissue-specific, reflecting the cell-type specific responses to elevated nutrient and energy status.

Muscle tissue is the major site of postprandial glucose disposal, and energy expenditure [16,17]. Muscle specific inhibition of mTORC1 (via ablation of *Rptor*), leads to weakened and short-lived mice [18,19]. Constitutive activation of mTORC1, via muscle-specific deletion of its negative regulator *Tsc1*, have identified age-related myoatrophy and a switch towards oxidative fiber types [20–22]. Consistent with this, studies in cell culture models implicate mTORC1 as also a positive regulator of mitochondrial function and ATP production [23–25]. mTORC1 is strongly activated in co-ordination with high protein diets or supplementation with essential amino acids [26,27]. We therefore hypothesized that in oxidative tissues such as heart and skeletal muscle, mTORC1 may promote increases in energy expenditure.

# Methods and Materials

## Animal Husbandry

All mice were purchased from The Jackson Laboratory. Unless otherwise stated, animals were fed a normal chow diet from Harlan Teklad. For High Fat Diet studies, animals were provided *ad libitum access* to a diet with 45% of calories from lard (Research Diets D1492).

For muscle specific knockouts, FVB-Tg(Ckmm-cre)5Khn/J transgenic mice (stock 006405) were crossed with floxed *Tsc1tm1Djk*/J mice (stock 005680). To generate F1 mice that were heterozygous for the floxed allele, and either had or lacked the *Ckmm-Cre* transgene. These parents were intercrossed to generate knockout mice (*Tsc1fl*/fl, *Ckmm-Cre*Tg/+), wild type mice(*Tsc1+/+*, *Ckmm-Cre*+/+) and controls containing the transgene only (*Tsc1+/+*, *Ckmm-Cre*Tg/+)or the floxed allele only (*Tsc1fl*/fl, *Ckmm-Cre*+/+). All four genotypes were evaluated for all experiments. If there were no significant differences between the three control strains, these were combined and labeled as Controls. Animals were sacrificed in either the fed or fasted state as indicated in the figure legends after isoflurane-induced anaesthesia and cervical dislocation at approximately ZT3. The University of Michigan and UTHSC Institutional Animal Care and Use Committees approved all animal procedures.

## Food Intake and Body Composition

Food intake was determined throughout the CD/HPD feeding studies by weighing the food in the cages (3 mice per cage) throughout the study and calculated based on the caloric content of the food. Food intake was therefore the average food eaten by a cage of mice divided by the number of mice in that cage. Body composition was determined using an echoMRI 1100 at approximately ZT10. Body weights were determined using a standard scale. Tissue weights were determined for both the left and right hand side tissue and combined as total weights.

## Energy Expenditure Studies

For high protein diet studies, physical movement and calorimetry was determined using a comprehensive laboratory animal monitoring system from Columbus Instruments. These experiments were performed in light and temperature controlled enclosures at 25C, in home-cage style cages with hanging feeders. The first 6h of measurements were discarded, after which animals were acclimatized to their new surroundings. Single-animal measurements were collected over the course of 3 days. Oxygen consumption was normalized to lean body mass (as determined by echoMRI, described above) and analyzed by mixed linear models with the considerations described in [28].

For rapamycin treatment, animals were injected with 3 mg/kg of rapamycin or a vehicle (1% Tween, 1% PEG-8000). For acute studies, animals were injected daily before switching diet to a high fat diet.

## Western Blotting

Protein lysates were generated in RIPA buffer (50 mM Tris pH 7.4, 0.25% sodium deoxycholate, 1% NP40, 150 mM sodium choride, 1 mM EDTA, 100 μM sodium vanadate, 5mM sodium fluoride, 10 mM sodium pyrophosphate and 1X protease inhibitors) or HTNG buffer (50 mM HEPES, pH 7.4, 150 mM sodium chloride, 10% glycerol, 10% triton X-100 and 1X protease inhibitors) by mechanical disruption in a Qialyser for 5 minutes at 30Hz. Lysates were clarified at 14 000 RPM for 15 minutes and quantified by Bradford assays. For PTG determination, glycogen binding proteins were enriched by amylose pull-down as previously described [11,29,30]. Proteins were separated by SDS-PAGE and blotted with antibodies described in the figure legends. Antibodies used in this study were raised against pS6K (pThr389, Cell Signaling cat #9206), S6K (Cell Signaling #2708), pAkt (pSer473, Cell Signaling #4060) and Akt (Cell Signaling #2920), TSC1 (Cell Signaling #4963), TSC2 (Cell Signaling #4308), pS6 (pSer236/236, Cell Signaling #2211), S6 (Cell Signaling #2317), SREBP1 (Santa Cruz #sc-366), pGS (pSer641, Cell Signaling #3891), GS (Cell Signaling #04-357) and PTG [11]. Blots were visualized via ECL or on a LiCor Odyssey and quantified using Image Studio Lite software.

## Triglyceride and Glycogen Determination

## Statistical Analyses

All statistical analyses were performed using the R, version 3.2.2 [31]. For longitudinal measurements (body weights, fat mass and lean mass), the data were analyzed by mixed linear models using uncorrelated random slopes and intercepts using the lme4 package version 1.1-8 [32]. Insulin tolerance tests were analyzed similarly with each time point treated as an repeated measurement. Statistical significance was determined via χ2 tests between models containing or missing the genotype or diet term. Pairwise comparisons were tested first for normality via a Shapiro-Wilk test, then for equal variance via Levene’s test. Based on these, appropriate pairwise tests were performed as indicated in the figure legends. Corrections for testing of multiple hypotheses were done using the method of Benjamini and Hochberg [33]. Statistical significance was designated at p/q<0.05 for all assays, except GSEA analyses were q<0.25 was used. All raw data and statistical analyses for this manuscript are available at <http://bridgeslab.github.io/TissueSpecificTscKnockouts>.

# Results

## Rapamycin Treatment Reduces High Fat Diet Induced Increases in Energy Expenditure

Both short term overfeeding and chronic obesity result in increased energy expenditure. To test whether mTORC1 plays a role in the short-term responses to obesegenic diets, we measured energy expenditure in single-housed mice during this dietary shift. As described in Figure 2A, animals were single housed and vehicle-injected daily for 3 days to acclimatize the animals. They were then placed in metabolic monitoring cages for 2 days, then were randomized into vehicle or rapamycin injected groups. After 2 days of these injections, all animals were moved from normal chow diets to a high fat diet (HFD). As shown in Figure 2B-C, this caused an increase in energy expenditure in the vehicle injected but not rapamycin treated animals. This was not associated with differences in physical activity as measured within these cages (Figures 2D-E). These data support the hypothesis that mTORC1 is required for the increase in energy expenditure in response to adaptation to HFD feeding. Together with the reduced adiposity and mTORC1-activating effects of high protein diets, we investigated a mouse model of mTORC1 activation, specifically in muscle tissues to test whether muscle mTORC1 activation was sufficient to affect adiposity and energy expenditure.

## Muscle *Tsc1* Deletion Elevates Fasting Glycogen and Triglyceride Levels

To test whether activation of mTORC1 in muscle caused the reduced fat mass observed in the high protein diet fed animals, we generated muscle specific *Tsc1* knockout mice using a floxed *Tsc1* allele [34] and the *Ckmm-Cre* transgene (muscle creatine kinase; expressed in both cardiac and skeletal muscle [35]). This driver differs from the *ACTA1-Cre* driven *Tsc1* knockout models previously described [20,21]. As shown in Figure 3A, we observed efficient knockout of TSC1 and TSC2 proteins, corresponding to an increase in mTORC1 activity in quadriceps lysates from these animals as determined by an increase in S6 phosphorylation (Figure 3B).

We next evaluated the cell autonomous changes in muscle tissue associated with *Tsc1* ablation. We found that both triglycerides and fasting glycogen levels were substantially elevated in *Tsc1* knockout muscles from fasted mice (Figures 3C-D). We repeated these studies in both fed and fasted muscle *Tsc1* knockout animals and found that this increase is only apparent in the fasted state (Supplementary Figure 2). The increase in fasting glycogen was correlated with increases in both processed SREPB1c (42% increase p=0.052), PTG protein levels and a decrease in the phosphorylation of GS (51% decrease, p=0.0012; Figure 3A-B). PTG has previously been shown to anchor protein phosphatase activity to the glycogen pellet [11,36], and the dramatic increase in muscle PTG protein is consistent with reduced GS phosphorylation and higher glycogen levels in these *Tsc1* knockout muscles. Mechanistically, our findings in muscle are consistent with our previous report of mTORC1-dependent, SREBP1c co-activation of the PTG promoter in the liver and *Tsc2* knockout embryonic fibroblasts [11].

## Deletion of *Tsc1* in Muscle Results in Reduced Fat Mass and Increased Energy Expenditure

We followed the body weights and composition of the muscle *Tsc1* knockout animals over the course of XX months. While we did not observe any differences in lean mass, we did observe a striking lack of fat mass accumulation as these animals grew in size on a normal chow diet, becoming more apparent as the animals aged (Figures 4A-C). Previous studies using *ACTA1-Cre* mediated knockout of *Tsc1* also observed reductions in fat mass, but in those cases lean mass was reduced as well [21,22]. To determine any adipose depot-specific changes, we measured the weights of fat pads from wild-type and *Tsc1* knockout mice, and found that both subcutaneous and epdidymal fat pads were reduced in size (Figures 4D-E).

To identify the cause of these reductions, we performed indirect calorimetry studies on these mice, prior to differences in adiposity (90 days of age).

## Ablation of Muscle *Tsc1* Results in Oxidative Muscle Fibers and Upregulation of Fatty Acid/Amino Acid Uptake Genes

## *Ckmm-Cre* driven knockout of *Tsc1* Impairs Glucose Disposal but Does Not Cause Insulin Resistance *in vivo*

## Knockout of *Tsc1* in Muscle Causes Resistance to Diet-Induced Obesity and Insulin Resistance

# Discussion

We report here that activation of mTORC1 in muscle tissue results in the accumulation of glycogen and triglycerides, with a shift towards more oxidative muscle fiber types. This, coupled with no detectable decrease in *in vivo* insulin sensitivity is reminiscent of the “athlete’s paradox”, in which muscles of endurance trained athletes are insulin normosensitive, in spite of increased nutrient deposition [37,38]. These findings are concordant with a role of mTORC1 in this process, as this kinase is also activated during resistance exercise and muscle growth [39,40].

Clinical studies have shown that energy expenditure can rapidly increase in response to overfeeding [3,41], consistent with the observation that obese individuals have higher energy expenditure than lean subjects [3,42–44]. Similarly, weight loss results in a rapid reduction in energy expenditure [2,3,6]. This compensation, when combined with increased feelings of hunger after weight loss [4], contributes to body weight set point maintenance and make successful, long term weight loss very difficult. Recent studies have shown that low carbohydrate (and high protein) diets are more successful in maintaining weight reductions [45], and that high protein diets also result in smaller reductions in energy expenditure [6]. A comparison of low-carbohydrate and low-fat calorie restricted diets (with equal protein intake) showed similar reductions of energy expenditure, consistent with a key role for protein in maintaining elevated energy expenditure during weight loss [46]. Similarly, overfeeding studies have shown that over-feeding induced increases in energy expenditure are even more pronounced when the diets are high in protein [5,47]. It is unlikely that brown adipose tissue mediates these effects as there is no increase in brown adipose tissue oxygen consumption in response to HFD [48], nor is HFD-induced energy expenditure impaired in *Ucp1* knockout mice [49].

Our identification of skeletal muscle mTORC1 as an anti-obesegenic provides a potential mechanism by which over-feeding can cause increases in energy expenditure. Our findings are consistent with the hypothesis that nutrient excess generally, and high protein feeding specifically may function via mTORC1-dependent changes in muscle oxidation, but do not preclude the role of other tissues and molecular targets that may underlie the increases in energy expenditure that accompany high protein diets (reviewed in [50]). While the direct mechanisms by which mTORC1 increases energy expenditure have not yet been identified, our data are congruent with previous reports showing that activation of mTORC1 in muscle results in elevated mitochondrial biogenesis in fibroblasts [51] and an increase in oxidative muscle fibers [20]. Our findings of reduced diet-induced obesity and increased energy expenditure are also generally congruent with the findings of Guridi *et al.* [22]*,* however our model has several important differences. We do not observe insulin resistance in our animals, nor do we observe myoatrophy as measured either by reduced lean body mass or increased expression of atrogenes. Furthermore, we do not observe transcriptional elevation of fatty acid synthase (*Fasn*) or GLUT1 (*Slc2a1*). Whether muscle atrophy, insulin resistance and the expression of these genes are linked is unclear, but these differences may reflect different Cre drivers used, as *ACTA1-Cre* was used in those studies, while *Ckmm-Cre* was used in our studies.

While one mechanism linking muscle mTORC1 activity to reduced fat mass may be elevations in muscle-derived FGF21 [22,52,53], another possibility is that these effects may be autonomous to the muscle tissue. The mechanism underlying the changes in fiber type are unclear but since amino acids and fatty acids are both reliant on mitochondria to generate fuel, it is possible that an alternation in substrate flux could result more oxidative fibers, which then leads to more increased lipid oxidation and less adiposity. This hypothesis is consistent with anti-obesegenic effects of muscle specific *Cd36* overexpression [54] and with the observation that elevated free fatty acids can promote mitochondrial biogenesis [55]. Increased nutrient uptake, coupled with elevated mitochondria would be likely to produce more ATP in these cells. This in and of itself would not be sufficient to produce heat, so we also propose that increased uncoupling of SERCA2 by sarcolipin may consume ATP to generate heat [56,57]. This would be consistent with upregulation of sarcolipin by HFD and exacerbated obesity when *Sln* is ablated [56,58]. As such muscle *Tsc1* knockout animals could be plausibly thermogenic due to multiple muscle-autonomous adaptations.

While further studies are needed to identify the roles of nutrient uptake, mitochondrial biogenesis and ATP utilization in mTORC1-dependent diet-induced thermogenesis, these findings support the hypothesis that activation of mTORC1 or its downstream targets, specifically in muscle tissue may play a role in diet-induced thermogenesis and there for be effective targets for weight loss interventions.

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# References

[1] World Health Organization., 2013. Obesity and Overweight. http://www.who.int/mediacentre/factsheets/fs311/en/.

[2] Leibel, R.L., Hirsch, J., 1984. Diminished energy requirements in reduced-obese patients. Metabolism 33(2): 164–70, Doi: 10.1016/0026-0495(84)90130-6.

[3] Leibel, R.L., Rosenbaum, M., Hirsch, J., 1995. Changes in energy expenditure resulting from altered body weight. The New England Journal of Medicine 332(10): 621–8, Doi: 10.1056/NEJM199503093321001.

[4] Sumithran, P., Prendergast, L.A., Delbridge, E., Purcell, K., Shulkes, A., Kriketos, A., et al., 2011. Long-Term Persistence of Hormonal Adaptations to Weight Loss. New England Journal of Medicine 365(17): 1597–604, Doi: 10.1056/NEJMoa1105816.

[5] Bray, G.A., Smith, S.R., de Jonge, L., Xie, H., Rood, J., Martin, C.K., et al., 2012. Effect of dietary protein content on weight gain, energy expenditure, and body composition during overeating: a randomized controlled trial. JAMA : The Journal of the American Medical Association 307(1): 47–55, Doi: 10.1001/jama.2011.1918.

[6] Ebbeling, C.B., Swain, J.F., Feldman, H. a., Wong, W.W., Hachey, D.L., Garcia-Lago, E., et al., 2012. Effects of dietary composition on energy expenditure during weight-loss maintenance. JAMA : The Journal of the American Medical Association 307(24): 2627–34, Doi: 10.1001/jama.2012.6607.

[7] Efeyan, A., Comb, W.C., Sabatini, D.M., 2015. Nutrient-sensing mechanisms and pathways. Nature 517(7534): 302–10, Doi: 10.1038/nature14190.

[8] Yecies, J.L., Zhang, H.H., Menon, S., Liu, S., Yecies, D., Lipovsky, A.I., et al., 2011. Akt Stimulates Hepatic SREBP1c and Lipogenesis through Parallel mTORC1-Dependent and Independent Pathways. Cell Metabolism 14(1): 21–32, Doi: 10.1016/j.cmet.2011.06.002.

[9] Chakrabarti, P., English, T., Shi, J., Smas, C.M., Kandror, K. V., 2010. Mammalian target of rapamycin complex 1 suppresses lipolysis, stimulates lipogenesis, and promotes fat storage. Diabetes 59(4): 775–81, Doi: 10.2337/db09-1602.

[10] Li, S., Brown, M.S., Goldstein, J.L., 2010. Bifurcation of insulin signaling pathway in rat liver: mTORC1 required for stimulation of lipogenesis, but not inhibition of gluconeogenesis. Proceedings of the National Academy of Sciences of the United States of America 107(8): 3441–6, Doi: 10.1073/pnas.0914798107.

[11] Lu, B., Bridges, D., Yang, Y., Fisher, K., Cheng, A., Chang, L., et al., 2014. Metabolic crosstalk: molecular links between glycogen and lipid metabolism in obesity. Diabetes 63(9): 2935–48, Doi: 10.2337/db13-1531.

[12] Zhang, H.H., Huang, J., Düvel, K., Boback, B., Wu, S., Squillace, R.M., et al., 2009. Insulin stimulates adipogenesis through the Akt-TSC2-mTORC1 pathway. PloS One 4(7): e6189, Doi: 10.1371/journal.pone.0006189.

[13] Hatfield, I., Harvey, I., Yates, E.R., Redd, J.R., Reiter, L.T., Bridges, D., 2015. The role of TORC1 in muscle development in Drosophila. Scientific Reports 5: 9676, Doi: 10.1038/srep09676.

[14] Erbay, E., Chen, J., 2001. The mammalian target of rapamycin regulates C2C12 myogenesis via a kinase-independent mechanism. The Journal of Biological Chemistry 276(39): 36079–82, Doi: 10.1074/jbc.C100406200.

[15] Shah, O.J., Wang, Z., Hunter, T., 2004. Inappropriate activation of the TSC/Rheb/mTOR/S6K cassette induces IRS1/2 depletion, insulin resistance, and cell survival deficiencies. Current Biology 14(18): 1650–6, Doi: 10.1016/j.cub.2004.08.026.

[16] DeFronzo, R.A., Ferrannini, E., Sato, Y., Felig, P., Wahren, J., 1981. Synergistic interaction between exercise and insulin on peripheral glucose uptake. The Journal of Clinical Investigation 68(6): 1468–74.

[17] Rolfe, D.F., Brown, G.C., 1997. Cellular energy utilization and molecular origin of standard metabolic rate in mammals. Physiological Reviews 77(3): 731–58.

[18] Bentzinger, C.F., Romanino, K., Cloëtta, D., Lin, S., Mascarenhas, J.B., Oliveri, F., et al., 2008. Skeletal muscle-specific ablation of raptor, but not of rictor, causes metabolic changes and results in muscle dystrophy. Cell Metabolism 8(5): 411–24, Doi: 10.1016/j.cmet.2008.10.002.

[19] Risson, V., Mazelin, L., Roceri, M., Sanchez, H., Moncollin, V., Corneloup, C., et al., 2009. Muscle inactivation of mTOR causes metabolic and dystrophin defects leading to severe myopathy. The Journal of Cell Biology 187(6): 859–74, Doi: 10.1083/jcb.200903131.

[20] Bentzinger, C.F., Lin, S., Romanino, K., Castets, P., Guridi, M., Summermatter, S., et al., 2013. Differential response of skeletal muscles to mTORC1 signaling during atrophy and hypertrophy. Skeletal Muscle 3(1): 6, Doi: 10.1186/2044-5040-3-6.

[21] Castets, P., Lin, S., Rion, N., Di Fulvio, S., Romanino, K., Guridi, M., et al., 2013. Sustained activation of mTORC1 in skeletal muscle inhibits constitutive and starvation-induced autophagy and causes a severe, late-onset myopathy. Cell Metabolism 17(5): 731–44, Doi: 10.1016/j.cmet.2013.03.015.

[22] Guridi, M., Tintignac, L.A., Lin, S., Kupr, B., Castets, P., Rüegg, M.A., 2015. Activation of mTORC1 in skeletal muscle regulates whole-body metabolism through FGF21. Science Signaling 8(402): ra113–ra113, Doi: 10.1126/scisignal.aab3715.

[23] Cunningham, J.T., Rodgers, J.T., Arlow, D.H., Vazquez, F., Mootha, V.K., Puigserver, P., 2007. mTOR controls mitochondrial oxidative function through a YY1-PGC-1alpha transcriptional complex. Nature 450(7170): 736–40, Doi: 10.1038/nature06322.

[24] Ramanathan, A., Schreiber, S.L., 2009. Direct control of mitochondrial function by mTOR. Proceedings of the National Academy of Sciences of the United States of America 106(52): 22229–32, Doi: 10.1073/pnas.0912074106.

[25] Koyanagi, M., Asahara, S.-I., Matsuda, T., Hashimoto, N., Shigeyama, Y., Shibutani, Y., et al., 2011. Ablation of TSC2 enhances insulin secretion by increasing the number of mitochondria through activation of mTORC1. PLoS ONE 6(8), Doi: 10.1371/journal.pone.0023238.

[26] Fujita, S., Dreyer, H.C., Drummond, M.J., Glynn, E.L., Cadenas, J.G., Yoshizawa, F., et al., 2007. Nutrient signalling in the regulation of human muscle protein synthesis. The Journal of Physiology 582(Pt 2): 813–23, Doi: 10.1113/jphysiol.2007.134593.

[27] Cuthbertson, D., Smith, K., Babraj, J., Leese, G., Waddell, T., Atherton, P., et al., 2005. Anabolic signaling deficits underlie amino acid resistance of wasting, aging muscle. The FASEB Journal : Official Publication of the Federation of American Societies for Experimental Biology 19(3): 422–4, Doi: 10.1096/fj.04-2640fje.

[28] Tschöp, M.H., Speakman, J.R., Arch, J.R.S., Auwerx, J., Brüning, J.C., Chan, L., et al., 2011. A guide to analysis of mouse energy metabolism. Nature Methods 9(1): 57–63, Doi: 10.1038/nmeth.1806.

[29] Cheng, A., Zhang, M., Gentry, M.S., Worby, C.A., Dixon, J.E., Saltiel, A.R., 2007. A role for AGL ubiquitination in the glycogen storage disorders of Lafora and Cori’s disease. Genes & Development 21(19): 2399–409, Doi: 10.1101/gad.1553207.

[30] Cheng, A., Zhang, M., Okubo, M., Omichi, K., Saltiel, A.R., 2009. Distinct mutations in the glycogen debranching enzyme found in glycogen storage disease type III lead to impairment in diverse cellular functions. Human Molecular Genetics 18(11): 2045–52, Doi: 10.1093/hmg/ddp128.

[31] R Core Team., 2013. R: A Language and Environment for Statistical Computing.

[32] Bates, D., Mächler, M., Bolker, B., Walker, S., 2014. Fitting Linear Mixed-Effects Models using lme4. ArXiv 1406.5823: 1–51.

[33] Benjamini, Y., Hochberg, Y., 1995. Controlling the False Discovery Rate: A Practical and Powerful Approach to Multiple Testing. Journal of the Royal Statistical Society. Series B 57(1): 289–300.

[34] Kwiatkowski, D.J., Zhang, H., Bandura, J.L., Heiberger, K.M., Glogauer, M., el-Hashemite, N., et al., 2002. A mouse model of TSC1 reveals sex-dependent lethality from liver hemangiomas, and up-regulation of p70S6 kinase activity in Tsc1 null cells. Human Molecular Genetics 11(5): 525–34.

[35] Brüning, J.C., Michael, M.D.D., Winnay, J.N., Hayashi, T., Hörsch, D., Accili, D., et al., 1998. A muscle-specific insulin receptor knockout exhibits features of the metabolic syndrome of NIDDM without altering glucose tolerance. Molecular Cell 2(5): 559–69, Doi: 10.1016/S1097-2765(00)80155-0.

[36] Printen, J.A., Brady, M.J., Saltiel, A.R., 1997. PTG, a protein phosphatase 1-binding protein with a role in glycogen metabolism. Science 275(5305): 1475–8.

[37] Goodpaster, B.H., He, J., Watkins, S., Kelley, D.E., 2001. Skeletal muscle lipid content and insulin resistance: Evidence for a paradox in endurance-trained athletes. Journal of Clinical Endocrinology and Metabolism 86(12): 5755–61, Doi: 10.1210/jc.86.12.5755.

[38] Amati, F., Dubé, J.J., Alvarez-Carnero, E., Edreira, M.M., Chomentowski, P., Coen, P.M., et al., 2011. Skeletal muscle triglycerides, diacylglycerols, and ceramides in insulin resistance: Another paradox in endurance-trained athletes? Diabetes 60(10): 2588–97, Doi: 10.2337/db10-1221.

[39] Philp, A., Hamilton, D.L., Baar, K., 2011. Signals mediating skeletal muscle remodeling by resistance exercise: PI3-kinase independent activation of mTORC1. Journal of Applied Physiology 110(2): 561–8, Doi: 10.1152/japplphysiol.00941.2010.

[40] Hamilton, D.L., Philp, A., MacKenzie, M.G., Patton, A., Towler, M.C.M.C., Gallagher, I.J., et al., 2014. Molecular brakes regulating mTORC1 activation in skeletal muscle following synergist ablation. American Journal of Physiology - Endocrinology And Metabolism 307(4): E365–73, Doi: 10.1152/ajpendo.00674.2013.

[41] Acheson, K.J., Schutz, Y., Bessard, T., Anantharaman, K., Flatt, J.P., Jequier, E., 1988. Glycoprotein storage capacity and de novo lipogenesis during massive carbohydrate overfeeding in man. American Journal of Clinical Nutrition 48(2): 240–7.

[42] Ravussin, E., Burnand, B., Schutz, Y., Jequier, E., 1982. Twenty-four-hour energy expenditure and resting metabolic rate in obese, moderately obese, and control subjects. American Journal of Clinical Nutrition 35(3): 566–73.

[43] Ravussin, E., Lillioja, S., Anderson, T.E., Christin, L., Bogardus, C., 1986. Determinants of 24-hour energy expenditure in man. Methods and results using a respiratory chamber. The Journal of Clinical Investigation 78(6): 1568–78, Doi: 10.1172/JCI112749.

[44] DeLany, J.P., Kelley, D.E., Hames, K.C., Jakicic, J.M., Goodpaster, B.H., 2013. High energy expenditure masks low physical activity in obesity. International Journal of Obesity (2005) 37(7): 1006–11, Doi: 10.1038/ijo.2012.172.

[45] Foster, G.D., Wyatt, H.R., Hill, J.O., Mcguckin, B.G., Brill, C., Mohammed, B.S., et al., 2003. A randomized trial of a low-carbohydrate diet for obesity. The New England Journal of Medicine 348(21): 2082–90, Doi: 10.1056/NEJMoa022207.

[46] Hall, K.D., Bemis, T., Brychta, R.J., Chen, K.Y., Courville, A., Crayner, E.J., et al., 2015. Calorie for Calorie, Dietary Fat Restriction Results in More Body Fat Loss than Carbohydrate Restriction in People with Obesity. Cell Metabolism 22(3): 427–36, Doi: 10.1016/j.cmet.2015.07.021.

[47] Bray, G.A., Redman, L.M., de Jonge, L., Covington, J.D., Rood, J., Brock, C., et al., 2015. Effect of protein overfeeding on energy expenditure measured in a metabolic chamber. American Journal of Clinical Nutrition 101(3): 496–505, Doi: 10.3945/ajcn.114.091769.

[48] Ma, S.W., Foster, D.O., Nadeau, B.E., Triandafillou, J., 1988. Absence of increased oxygen consumption in brown adipose tissue of rats exhibiting “cafeteria” diet-induced thermogenesis. Canadian Journal of Physiology and Pharmacology 66(11): 1347–54.

[49] Anunciado-Koza, R., Ukropec, J., Koza, R. a., Kozak, L.P., 2008. Inactivation of UCP1 and the glycerol phosphate cycle synergistically increases energy expenditure to resist diet-induced obesity. The Journal of Biological Chemistry 283(41): 27688–97, Doi: 10.1074/jbc.M804268200.

[50] Halton, T.L., Hu, F.B., 2004. The effects of high protein diets on thermogenesis, satiety and weight loss: a critical review. Journal of the American College of Nutrition 23(5): 373–85, Doi: 10.1080/07315724.2004.10719381.

[51] Morita, M., Gravel, S.P., Chénard, V., Sikström, K., Zheng, L., Alain, T., et al., 2013. MTORC1 controls mitochondrial activity and biogenesis through 4E-BP-dependent translational regulation. Cell Metabolism 18(5): 698–711, Doi: 10.1016/j.cmet.2013.10.001.

[52] Tsai, S., Sitzmann, J.M., Dastidar, S.G., Rodriguez, A. a., Vu, S.L., McDonald, C.E., et al., 2015. Muscle-specific 4E-BP1 signaling activation improves metabolic parameters during aging and obesity. The Journal of Clinical Investigation 125(8): 2952–64, Doi: 10.1172/JCI77361.

[53] Kim, K.H., Jeong, Y.T., Oh, H., Kim, S.H., Cho, J.M., Kim, Y.-N., et al., 2012. Autophagy deficiency leads to protection from obesity and insulin resistance by inducing Fgf21 as a mitokine. Nature Medicine, Doi: 10.1038/nm.3014.

[54] Ibrahimi, A., Bonen, A., Blinn, W.D., Hajri, T., Li, X., Zhong, K., et al., 1999. Muscle-specific overexpression of FAT/CD36 enhances fatty acid oxidation by contracting muscle, reduces plasma triglycerides and fatty acids, and increases plasma glucose and insulin. Journal of Biological Chemistry 274(38): 26761–6, Doi: 10.1074/jbc.274.38.26761.

[55] Garcia-Roves, P., Huss, J.M., Han, D.-H., Hancock, C.R., Iglesias-Gutierrez, E., Chen, M., et al., 2007. Raising plasma fatty acid concentration induces increased biogenesis of mitochondria in skeletal muscle. Proceedings of the National Academy of Sciences of the United States of America 104(25): 10709–13, Doi: 10.1073/pnas.0704024104.

[56] Bal, N.C., Maurya, S.K., Sopariwala, D.H., Sahoo, S.K., Gupta, S.C., Shaikh, S.A., et al., 2012. Sarcolipin is a newly identified regulator of muscle-based thermogenesis in mammals. Nature Medicine 18(10): 1575–9, Doi: 10.1038/nm.2897.

[57] Sahoo, S.K., Shaikh, S.A., Sopariwala, D.H., Bal, N.C., Periasamy, M., 2013. Sarcolipin Protein Interaction with Sarco(endo)plasmic Reticulum Ca2+ATPase (SERCA) Is Distinct from Phospholamban Protein, and Only Sarcolipin Can Promote Uncoupling of the SERCA Pump. Journal of Biological Chemistry 288(10): 6881–9, Doi: 10.1074/jbc.M112.436915.

[58] Bombardier, E., Smith, I.C., Gamu, D., Fajardo, V.A., Vigna, C., Sayer, R.A., et al., 2013. Sarcolipin trumps β-adrenergic receptor signaling as the favored mechanism for muscle-based diet-induced thermogenesis. FASEB Journal 27(9): 3871–8, Doi: 10.1096/fj.13-230631.

# Figure Legends

**Figure 1: Effects of high protein diet on body composition and energy expenditure.** Body composition was determined weekly with body weight (A), lean mass (B) and fat mass (C). D) Weights of fat pad depots at the end of the 14 week feedint study. Statistical significance (p<0.05, n=12/group) was denoted via asterisks based on χ2 test (A-C) or Student’s *t-*test (D).

**Figure 2: Regulation of mTORC1 by amino acids and protein feeding.** A) XXXXXX C) Western blotting of quadriceps RIPA lysates from 16h fasted or non-fasted animals D) Quantification of the blots in B.

**Figure 3: Knockout of *Tsc1* in muscle leads to increased fasted glycogen and triglyceride levels in quadriceps.** A)Protein lysates from quadriceps dissected from animals fasted overnight were prepared in HNTG buffer and blotted as indicated. B) Quantification of processed SREBP1 and the phosphorylation ratio of glycogen synthase. Statistical significance (p<0.05, n=5/7) was determined via a Wilcoxon Rank-Sum test (C) after correcting for multiple hypotheses.

# Supplementary Figure Legends

**Supplementary Table 1: Gene expression differences in muscle *Tsc1* knockout quadriceps.**