TABLE 1. Channel kinetics and functional K_{ATP} channel density at the cell surface

K_{ATP} channel	Open probability (Po)	Conductance (pS)	Channel density	
			Rate of detectable channels (%)	Channel number in a patch
Kir6.2/SUR1	0.13 ± 0.01	78.6 ± 3.2	54.8	8.35 ± 1.10
Kir6.2 C42R/SUR1	0.57 ± 0.04[a]	80.9 ± 3.0	23.3[a]	1.50 ± 0.25[a]

Recordings were made at −70 mV in ATP-free conditions (n = 4 for the open probability, n = 6 for the channel conductance, n = 60–62 for the rate of detectable channels, n = 34 for Kir6.2/SUR1, and n = 14 for Kir6.2 C42B/SUR1 for the number of channels in patch determination).
[a] $P < 0.01$.

comparable with the mutations reported in patients with milder permanent neonatal diabetes (14) but that these diabetogenic effects were partially compensated by the decreased expression of the mutant Kir6.2 at the cell surface. This reduction in expression probably accounts for the reason that our patients showed relatively milder phenotypes and later onset rather than PND. The molecular mechanism leading to the reduction in expression is currently unknown. One possibility is that the C42R mutation affected coassembly of the mutant Kir6.2 with the SUR1 subunits because it has been shown that the coassembly is important for the effective sorting of the channel to the cell surface (14).

Other than the mutations in the KCNJ11 gene that cause the extremely rare PND cases, more common single-nucleotide polymorphisms (SNPs) in the same gene, such as E23K, have been shown to confer a predisposition to type 2 diabetes, with an odds ratio of 1.18–1.49 (4, 8). Although much milder, the biochemical basis of this type of diabetes is basically the same as for PND: reduced ATP-sensitivity and increased open probabilities (6, 9, 15). Taken together with our results, these results reveal the phenotypic spectrum of diabetes caused by the mutated KCNJ11, the mildest end being common SNPs such as E23K, which is a risk factor for type 2 diabetes (its effect is evident only in large-scale association studies), and the most severe end being the extremely rare PND mutations with very high penetrance. The in-between mutations, such as C42R, cause milder diabetes but with much higher penetrance than the common SNPs. The exact prevalence of these moderate mutations in KCNJ11 is currently under investigation. We have already searched for the KCNJ11 mutations in five additional Japanese families with similar clinical characteristics. At present, none had mutations in KCNJ11, probably suggesting that this gene is not the major unidentified MODY gene in the Japanese population.

Clinically it is important to identify patients with mutations in KCNJ11 because it has been shown that oral sulfonylurea might be effective for treating even severely affected patients (9, 16). Two of our patients (II-3, III-1) could be successfully treated with oral sulfonylurea alone. Although tolbutamide sensitivity of the C42R mutation was reduced as assayed in the homozygous state, the reduction in sensitivity could be much milder when assayed in the heterozygous state, *i.e.* cotransfection of the wild-type and mutant Kir6.2. This phenomenon has previously been reported for the R210H mutation (9) and could be an explanation for this apparent discrepancy between the sensitivity data and the clinical responses.

The severity of diabetes caused by mutations in the KCNJ11 gene probably reflects the variable effects of each mutation on the Kir6.2 molecules, which includes ATP sensitivity, spontaneous Po, sulfonylurea sensitivity, or cell surface expression. In addition, the presented cases demonstrate that, with the same mutation, the clinical presentation could be variable. Moreover, even in the same patient, the disease severity fluctuates from time to time. This suggests that other factors, both genetic and environmental, affect the clinical presentation of the patients with milder KCNJ11 mutations. Although the efficacy of sulfonylurea therapy appears promising, it could also be affected by the above factors. Accumulation of more cases is necessary to achieve the optimal management of these patients.

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Tacrolimus suppresses glucose-induced insulin release from pancreatic islets by reducing glucokinase activity

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Radu, Razvan Gheorghe, Shimpei Fujimoto, Eri Mukai, Mihoko Takehiro, Dai Shimono, Koichiro Nabe, Makiko Shimodahira, Rieko Kominato, Yo Aramaki, Yuichi Nishi, Shogo Funakoshi, Yuichiro Yamada, and Yutaka Seino. Tacrolimus suppresses glucose-induced insulin release from pancreatic islets by reducing glucokinase activity. *Am J Physiol Endocrinol Metab* 288: E365–E371, 2005. First published October 12, 2004; doi:10.1152/ajpendo.00390.2004.—Tacrolimus is widely used for immunosuppressant therapy, including various organ transplantations. One of its main side effects is hyperglycemia due to reduced insulin secretion, but the mechanism remains unknown. We have investigated the metabolic effects of tacrolimus on insulin secretion at a concentration that does not influence insulin content. Twenty-four-hour exposure to 3 nM tacrolimus reduced high glucose (16.7 mM)-induced insulin secretion (control 2.14 ± 0.08 vs. tacrolimus 1.75 ± 0.02 ng·islet^{-1}·30 min^{-1}, $P < 0.01$) without affecting insulin content. In dynamic experiments, insulin secretion and NAD(P)H fluorescence during a 20-min period after 10 min of high-glucose exposure were reduced in tacrolimus-treated islets. ATP content and glucose utilization of tacrolimus-treated islets in the presence of 16.7 mM glucose were less than in control (ATP content: control 9.69 ± 0.99 vs. tacrolimus 6.52 ± 0.40 pmol/islet, $P < 0.01$; glucose utilization: control 103.8 ± 6.9 vs. tacrolimus 74.4 ± 5.1 pmol·islet^{-1}·90 min^{-1}, $P < 0.01$). However, insulin release from tacrolimus-treated islets was similar to that from control islets in the presence of 16.7 mM α-ketoisocaproate, a mitochondrial fuel. Glucokinase activity, which determines glycolytic velocity, was reduced by tacrolimus treatment (control 65.3 ± 3.4 vs. tacrolimus 49.9 ± 2.8 pmol·islet^{-1}·60 min^{-1}, $P < 0.01$), whereas hexokinase activity was not affected. These results indicate that glucose-stimulated insulin release is decreased by chronic exposure to tacrolimus due to reduced ATP production and glycolysis derived from reduced glucokinase activity.

islet; adenosine 5′-triphosphate

TACROLIMUS (FK-506) IS AN IMMUNOSUPPRESSANT widely used in human organ transplantation. Immunosuppression by the agent is due to blocking of antigen-stimulated expression of genes, including interleukin-2 in T lymphocytes, which is required for T-cell proliferation (34). Interleukin-2 gene transcription is activated by dephosphorylation and nuclear translocation of a transcriptional cofactor, the nuclear factor of activated T cells (NFAT). Tacrolimus binds specific intracellular proteins, FK-506-binding proteins (FKBPs), and inhibits calcineurin (protein phosphatase-2B), a Ca^{2+}/calmodulin-dependent Ser/Thr phosphatase (7, 28) that dephosphorylates and translocates NFAT to nuclei (20, 37). Thus tacrolimus suppresses interleukin-2 gene transcription by inhibiting the calcineurin/NFAT pathway.

The main adverse reaction in tacrolimus-treated patients is hyperglycemia due to reduced insulin release (6, 16, 26). The mechanism of the inhibitory effect of tacrolimus on insulin release has been described. Tacrolimus markedly reduces the number of endocrine secretory granules in human pancreatic β-cells, which correlates with the blood concentration of the agent and is reversible by reduction of the dose (5, 15). The agent decreases insulin release, insulin content, and insulin mRNA concentration dependently in experiments using cell lines, rat islets, and human islets (27, 30, 31, 32). Tacrolimus also suppresses insulin gene expression, which is induced by intracellular Ca^{2+} elevation via the calcineurin/NFAT pathway (17). Thus reduced insulin content due to reduced insulin synthesis plays an important role in the inhibitory effect of tacrolimus on insulin release.

The mechanism of glucose-stimulated insulin release from pancreatic β-cells has been well documented. Glucose stimulates insulin secretion by both triggering and amplifying signals in pancreatic β-cells (14). The triggering pathway includes entry of glucose into β-cells, acceleration of glycolysis in cytosol and glucose oxidation in mitochondria, an increase in ATP content and ATP-to-ADP ratio, closure of ATP-sensitive K^+ (K_{ATP}) channels, membrane depolarization, opening of voltage-dependent Ca^{2+} channels (VDCCs), an increase in Ca^{2+} influx through VDCCs, raised intracellular Ca^{2+} concentration ($[Ca^{2+}]_i$), and exocytosis of insulin granules. It has been reported that glucose also enhances insulin secretion K_{ATP} channel independently. The K_{ATP} channel-independent amplifying action of glucose has been confirmed by treatment of β-cells with diazoxide, which prevents K_{ATP} channels from closing, and with a depolarizing concentration of extracellular K^+ that restores Ca^{2+} influx (1, 12). Because glucose does not increase $[Ca^{2+}]_i$ but nevertheless augments insulin release under these conditions, glucose may well exert its effects by increasing Ca^{2+} efficacy in stimulation-secretion coupling, due at least in part to the direct effect of increased ATP derived from glucose metabolism on exocytosis (14). Thus glucose metabolism in β-cells plays a crucial role in glucose-induced insulin release. However, the effect of tacrolimus on glucose-induced insulin release and glucose metabolism has not been examined in detail.

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We have investigated the effects of chronic exposure to tacrolimus on insulin release by functional analysis of the metabolic responses of tacrolimus-treated islets.

MATERIALS AND METHODS

Materials. Tacrolimus (FK-506) was donated by Fujisawa Pharmaceutical (Tokyo, Japan). RPMI 1640 and glucose-6-phosphate dehydrogenase (G6PDH) were obtained from Sigma (St. Louis, MO). Luciferin-luciferase was obtained from Turner Designs (Sunnyvale, CA). ^3H_2O and [5-^{3}H]glucose were obtained from Amersham (Buckinghamshire, UK). All other agents were obtained from Nacalai Tesque (Kyoto, Japan).

Animals. Male Wistar rats weighing 180–230 g were obtained from Shimizu (Kyoto, Japan). The animals were fed standard lab chow ad libitum and allowed free access to water in an air-conditioned room with a 12:12-h light-dark cycle until the experiments. All experiments were carried out with rats aged 8–12 wk. The animals were maintained and used in accordance with the Guidelines for Animal Experiments of Kyoto University.

Islet isolation and culture. Islets of Langerhans were isolated from Wistar rats by collagenase digestion, as described previously (8). Isolated islets were cultured for 24 h in RPMI 1640 medium containing 10% FCS, 100 U/ml penicillin, 100 μg/ml streptomycin, and 5.5 mM glucose with or without tacrolimus at 37°C in humidified air containing 5% CO_2.

Measurement of insulin release from isolated rat pancreatic islets. Insulin release from intact islets was monitored using either batch incubation or a perifusion system described previously, using Krebs-Ringer bicarbonate buffer (KRBB) supplemented with 0.2% bovine serum albumin (BSA; fraction V) and 10 mM HEPES, adjusted to pH 7.4 (KRBB medium) (8).

For static incubation experiments, cultured islets were preincubated at 37°C for 30 min in KRBB medium supplemented with 2.8 mM glucose. Groups of five islets were then batch incubated for 30 min in 0.7 ml of KRBB medium with test materials. At the end of the incubation period, the islets were pelleted by centrifugation, and aliquots of the buffer were sampled.

For perifusion experiments, groups of 20 cultured islets were placed in each of the parallel chambers of a perifusion apparatus and perifused with KRBB medium at a rate of 0.7 ml/min at 37°C. The medium was continuously gassed with 95% O_2-5% CO_2. The islets were perifused for 30 min to establish a stable insulin secretory rate at a basal level of glucose and then exposed to the stimulating levels of glucose.

The amount of immunoreactive insulin was determined by RIA with rat insulin as standard.

Measurement of insulin and DNA content. Insulin contents and DNA contents of islets were measured as previously described (10). Briefly, after an aliquot of incubation medium for insulin release assay was taken, the islets remaining were lysed. Insulin content was determined by RIA using diluted samples. DNA content was determined by fluorometric assay using bisbenzimidazole (Hoechst 33258) as fluorochrome and calf thymus DNA (type I, Sigma) as standard.

Measurement of NAD(P)H fluorescence. Cultured islets without dye were immediately placed in a heat-controlled chamber on the stage of an inverted microscope kept at 36 ± 1°C and superfused with KRBB medium with 2.8 mM glucose for 30 min. The islets were excited successively at 360 nm, and the fluorescence emitted at 470 nm (13) was captured every 20 s by a charge-coupled device camera (Micro Max 5-MHz System; Roper Industries, Trenton, NJ). Changes in NAD(P)H fluorescence signal were expressed as percent control values by dividing the signal at a given time by the average signal at 2.8 mM glucose during the last 5 min before stimulation. Images of NAD(P)H were analyzed with the Meta Fluor image analyzing system (Universal Imaging, West Chester, PA).

Measurement of ATP content. ATP contents were determined as previously described, with a slight modification (10). Briefly, after groups of cultured islets were preincubated at 2.8 mM glucose for 30 min, groups of 15 islets were batch incubated in 0.5 ml of KRBB medium containing 2.8 or 16.7 mM glucose at 37°C for 30 min. Incubation was stopped by the addition of 0.1 ml of 2 M $HClO_4$. The tubes were immediately mixed with vortex and sonicated in ice-cold water. They were then centrifuged, and a fraction (0.4 ml) of the supernatant was mixed with 0.1 ml of 2 M HEPES and 0.1 ml of 1 M Na_2CO_3. The ATP concentration was measured by adding 0.2 ml of luciferin-luciferase solution to a fraction sample (0.1 ml) in a bioluminometer (Luminometer model 20e; Turner Designs). To draw a standard curve, blanks and ATP standards were run through the entire procedure, including the extraction steps.

Measurement of glucose utilization. Glucose utilization was measured, using a previously described method (2) with a slight modification. Cultured islets were preincubated in KRBB medium with 2.8 mM glucose at 37°C for 30 min. Batches of 30 islets for each condition were incubated at 37°C for 90 min in 150 μl of medium containing 1.5 μCi of [5-^{3}H]glucose. Aliquots of the incubation medium (100 μl) and 20 μl of 1 M HCl were transferred into small tubes and placed into a glass vial containing 0.5 ml of H_2O. The capped vials were incubated overnight at 37°C to vaporize ^3H_2O from the solution. The inner tube was then lifted out, and the disintegrations per minute of water-melting ^3H_2O in the vial were counted. In a parallel incubation, the recovery ratio of ^3H_2O was measured using ^3H_2O. After subtracting blank disintegrations per minute from sample disintegrations per minute, glucose utilization was calculated using the disintegrations per minute, specific radioactivity of [5-^{3}H]glucose, and recovery ratio of ^3H_2O.

Measurement of glucokinase and hexokinase activity. Glucokinase activity was measured by a fluorometric assay according to a method reported previously (25, 38). After cultured islets were preincubated with KRBB medium with 2.8 mM glucose, 100 islets were homogenized in 250 μl of solution containing 20 mM KH_2PO_4, 100 mM KCl, 1 mM $MgCl_2$, 1 mM EDTA, 5% glycerol, and 1 mM DTT (pH 7 by KOH), and the supernatants (islet extracts) were obtained from the homogenates by centrifugation (10,000 *g*, 15 min) at 4°C. The glucose phosphorylation rate was estimated as the increase in NADH through the following reaction: glucose-6-phosphate + NAD → 6-phosphoglucono-δ-lactone + NADH by NAD-dependent G6PDH. The enzyme reaction was performed using 25 μl of islet extracts in a 160-μl solution consisting of 50 mM HEPES (pH 7.4 by NaOH), 100 mM KCl, 8 mM $MgCl_2$, 0.5 mM NAD, 5 mM ATP, 1 mM DTT, and 1 U/ml G-6-PDH supplemented with two concentrations (50 mM and 0.5 mM) of glucose at 37°C for 1 h; it was stopped by adding 290 μl of stopping solution (300 mM Na_2HPO_4, 0.46 mM SDS, pH 8.0). NADH concentration was measured by fluorometry (Shimazu RF-5000, Kyoto, Japan) at a 340-nm excitation and a 450-nm emission. Blanks in the absence of ATP were incubated in a parallel experiment and were subtracted from the total fluorescence of the corresponding complete reaction mixtures. Glucokinase activity was determined by subtracting hexokinase activity measured at 0.5 mM glucose from the activity measured at 50 mM glucose.

Statistical analysis. Results are expressed as means ± SE. Statistical significance was evaluated by unpaired Student's *t*-test. $P < 0.05$ was considered significant.

RESULTS

Effect of chronic exposure to tacrolimus on glucose-induced insulin release, insulin content, and DNA content in islets. Chronic (24 h) exposure to tacrolimus (3–30 nM) concentrations dependently suppressed 16.7 mM glucose-induced insulin release but did not affect basal insulin release in the presence of 2.8 mM glucose (Table 1). Insulin content was significantly reduced by 24-h exposure to 10 and 30 nM tacrolimus (Table 2). However, 24-h exposure to 3 nM tacroli-

Table 1. *Concentration-dependent effect of chronic exposure to tacrolimus on glucose-induced insulin release from pancreatic islets*

Experimental Condition During Culture	Insulin Release, ng·islet^{-1}·30 min^{-1}	
	Basal (2.8 mM glucose)	Stimulated (16.7 mM glucose)
Control	0.26±0.02	2.14±0.08
3 nM Tacrolimus	0.26±0.03	1.75±0.02*
10 nM Tacrolimus	0.24±0.02	1.53±0.04*
30 nM Tacrolimus	0.21±0.02	1.49±0.03*

Values represent means ± SE of 16 determinations from several experiments. Islets were cultured under the conditions indicated for 24 h. After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM glucose for 30 min was determined. *$P < 0.01$ vs. control (cultured without tacrolimus).

mus significantly suppressed high glucose-induced insulin release but did not affect insulin content (Tables 1 and 2). DNA content was not altered by 24-h exposure to 3, 10, and 30 nM tacrolimus, indicating that these concentrations of tacrolimus do not reduce islet mass (Table 2). Accordingly, islets cultured with 3 nM tacrolimus for 24 h were used to investigate the reduction in glucose-induced insulin release without the influence of altered insulin content. In perifusion experiments, basal insulin release was similar in control and tacrolimus-treated islets (Fig. 1*A*). High glucose-induced biphasic insulin release from tacrolimus-treated islets was reduced compared with control islets after 10 min of high-glucose exposure (Fig. 1*A*). Mean insulin release during the first 10 min after high-glucose exposure was not affected, whereas that during the second and third 10-min period was suppressed in tacrolimus-treated islets (mean insulin release from 20 to 30 min: tacrolimus 0.60 ± 0.03 vs. control 0.86 ± 0.06 ng·10 islets^{-1}·min^{-1}, $n = 6$, $P < 0.01$; Fig. 1*B*). Exposure (12 h) to 3 nM tacrolimus inhibited high glucose-induced insulin release to the level brought about by 24-h exposure. However, the inhibitory effect of 12-h exposure to 3 nM tacrolimus was almost completely recovered by the withdrawal of tacrolimus for 12 h (Table 3).

Effect of chronic exposure to tacrolimus on α-ketoisocaproate-induced insulin release. Inhibitors of glycolysis, including mannoheptulose (20 mM) and iodoacetate (1 mM), almost completely suppressed glucose-induced insulin release but did not affect α-ketoisocaproate (KIC)-induced insulin release (Table 4). These results indicate that KIC-induced insulin release is not affected by inhibition of glycolysis. To characterize metabolic fuel-induced insulin release independent of the inhibition of glycolysis, KIC-induced insulin release from tacrolimus-treated islets was examined. Chronic exposure to 3 nM tacrolimus did not affect KIC (16.7 mM)-induced insulin release (Table 5).

Table 2. *Effect of chronic exposure to tacrolimus on insulin content and DNA content*

Experimental Condition During Culture	Insulin Content, ng/islet	DNA Content, ng/islet
Control	25.0±0.7	19.3±0.7
3 nM Tacrolimus	26.2±1.0	21.1±0.8
10 nM Tacrolimus	22.4±1.1*	20.1±1.0
30 nM Tacrolimus	20.4±0.8*	19.9±1.0

Values represent means ± SE of 16 determinations from several experiments. At the end of the experiments indicated in Table 1, insulin content and DNA content were determined, using islets in a randomly selected half of the batches. *$P < 0.01$ vs. control (cultured without tacrolimus).

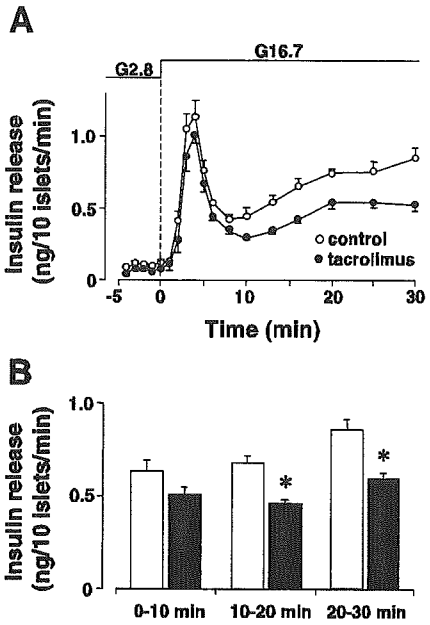

Fig. 1. High (16.7 mM) glucose-induced biphasic insulin release in control and tacrolimus-treated islets. Islets were cultured with or without 3 nM tacrolimus for 24 h. After cultured islets were perifused with 2.8 mM glucose (G2.8) for 30 min, they were stimulated with 16.7 mM glucose (G16.7) at *time 0* for 30 min. *A*: time course of high glucose-induced biphasic insulin release in control and tacrolimus-treated islets. Values represent means ± SE of 6 determinations. All values in tacrolimus-treated islets after 10 min of exposure to high glucose were significantly less than the corresponding values in control ($P < 0.01$). *B*: average values calculated from the data in *A*. Average values during indicated time period after high-glucose exposure in control (open bar) and tacrolimus-treated islets (solid bar) are shown. *$P < 0.01$ vs. corresponding control.

Time course of NAD(P)H fluorescence in islets. NAD(P)H fluorescence began to elevate immediately after exposure to

Table 3. *Reversible effect of tacrolimus on glucose-induced insulin release from islets*

Experimental Condition During Culture	Insulin Release, ng·islet^{-1}·30 min^{-1}	
	Basal (2.8 mM glucose)	Stimulated (16.7 mM glucose)
Control (24-h no tacrolimus)	0.26±0.02	2.15±0.09
24-h Tacrolimus	0.26±0.02	1.65±0.06*
12-h No tacrolimus + 12-h tacrolimus	0.24±0.01	1.72±0.07*
12-h Tacrolimus + 12-h no tacrolimus	0.28±0.03	2.24±0.09†

Values represent means ± SE of 8 determinations from several experiments. Two groups of islets were cultured with or without 3 nM tacrolimus for 24 h (control and 24-h tacrolimus group). One group of islets was cultured without tacrolimus for the first 12 h and with 30 nM tacrolimus for the second 12 h (12-h no tacrolimus + 12-h tacrolimus group). Another group of islets was cultured with 30 nM tacrolimus for the first 12 h and without tacrolimus for the second 12 h (12-h tacrolimus, + 12-h no tacrolimus group). After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM glucose for 30 min was determined. *$P < 0.01$ vs. control (24-h no tacrolimus). †$P < 0.01$ vs. 24-h tacrolimus. Stimulated insulin release was not different between 24-h tacrolimus and 12-h no tacrolimus + 12-h tacrolimus and between control (24-h no tacrolimus) and 12-h tacrolimus + 12-h no tacrolimus.

Table 4. *Effect of glycolytic inhibition on glucose-induced and KIC-induced insulin release from pancreatic islets*

	Insulin Release, ng·islet^{-1}·30 min^{-1}			
	Glucose		KIC	
Experimental Condition	2.8 mM	16.7 mM	2.8 mM	16.7 mM
Control	0.25±0.01	2.15±0.10	0.25±0.04	1.91±0.09
20 mM Mannoheptulose	0.22±0.03	0.24±0.02*	0.24±0.02	1.99±0.13
1 mM Iodoacetate	0.22±0.01	0.27±0.02*	0.23±0.01	2.02±0.08

Values represent means ± SE of 16 determinations from several experiments. Islets were cultured without tacrolimus for 24 h. After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM glucose or α-ketoisocaproate (KIC) for 30 min with or without glycolytic inhibitors was determined. *$P < 0.01$ vs. control (16.7 mM glucose without inhibitors).

high glucose in both groups of islets and reached a plateau within 10 min after exposure (Fig. 2*A*). A significant attenuation of NAD(P)H fluorescence was observed from 10 min in tacrolimus-cultured islets (at 10 min: tacrolimus 124.9 ± 1.3, $n = 11$, vs. control 129.3 ± 1.1%, $n = 12$, $P < 0.05$; Fig. 2*A*). Mean NAD(P)H fluorescence during the first 10 min after high-glucose exposure was not affected, whereas that during the second and third 10-min period was suppressed in tacrolimus-treated islets [mean NAD(P)H fluorescence from 20 to 30 min: tacrolimus 124.5 ± 1.2, $n = 11$, vs. control 133.9 ± 1.4%, $n = 12$, $P < 0.01$; Fig. 2*B*].

Effect of tacrolimus on ATP content. ATP content was greater in islets incubated with 16.7 mM glucose than in islets incubated with 2.8 mM glucose in both control (2.8 mM glucose, 5.22 ± 0.51, vs. 16.7 mM glucose, 9.69 ± 0.99 pmol/islet, $n = 10$, $P < 0.01$) and tacrolimus-treated islets (2.8 mM glucose, 4.75 ± 0.50, vs. 16.7 mM glucose, 6.52 ± 0.40 pmol/islet, $n = 10$, $P < 0.05$). ATP content in the presence of 16.7 mM glucose was significantly reduced in tacrolimus-treated islets ($P < 0.01$) but in the presence of 2.8 mM glucose was not affected by tacrolimus (Fig. 3*A*).

Effect of tacrolimus on glucose utilization. Glucose utilization was greater in islets incubated with 16.7 mM glucose than in islets incubated with 2.8 mM glucose in both control (2.8 mM glucose, 35.1 ± 6.1, vs. 16.7 mM glucose, 103.8 ± 6.9 pmol·islet^{-1}·90 min^{-1}, $n = 9$, $P < 0.01$) and tacrolimus-treated islets (2.8 mM glucose, 24.6 ± 3.7, vs. 16.7 mM glucose, 73.4 ± 5.1 pmol·islet^{-1}·90 min^{-1}, $n = 9$, $P < 0.01$). Glucose utilization in the presence of 16.7 mM glucose was significantly reduced in tacrolimus-treated islets ($P < 0.01$) but in the presence of 2.8 mM glucose was not affected by tacrolimus (Fig. 3*B*).

Table 5. *Effect of chronic exposure to tacrolimus on KIC-induced insulin release from pancreatic islets*

	Insulin Release, ng·islet^{-1}·30 min^{-1}	
Experimental Condition During Culture	Basal (2.8 mM KIC)	Stimulated (16.7 mM KIC)
Control	0.28±0.02	2.09±0.04
3 nM Tacrolimus	0.23±0.02	2.05±0.04

Values represent means ± SE of 16 determinations from several experiments. Islets were cultured under the conditions indicated for 24 h. After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM KIC for 30 min was determined.

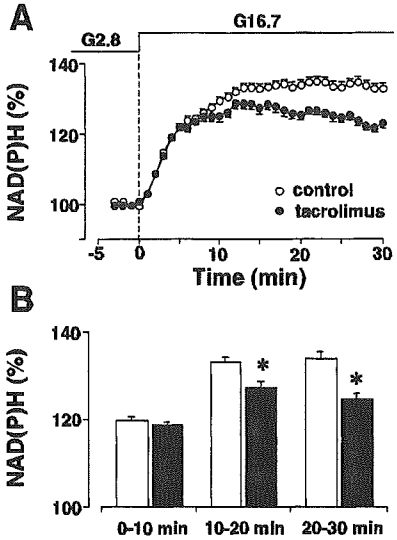

Fig. 2. High (16.7 mM) glucose-induced NAD(P)H fluorescence elevation in control and tacrolimus-treated islets. Islets were cultured with or without 3 nM tacrolimus for 24 h. After cultured islets were perifused with 2.8 mM glucose for 30 min, they were stimulated with 16.7 mM glucose at *time 0* for 30 min. *A*: time course of high glucose-induced NAD(P)H fluorescence elevation in control and tacrolimus-treated islets. Values represent means ± SE of 12 (control) or 11 (tacrolimus) determinations from the several experiments. All values, except those at 12 min, in tacrolimus-treated islets after 10 min of exposure to high glucose were significantly less than the corresponding values in control (at 10, 11, 13, 14, 15, and 16 min: $P < 0.05$; after 17 min: $P < 0.01$). *B*: average values calculated from the data in *A*. Average values during indicated time period after high-glucose exposure in control (open bar) and tacrolimus-treated islets (solid bar) are shown. *$P < 0.01$ vs. corresponding control.

Effect of tacrolimus on glucokinase and hexokinase activity. Glucokinase activity was significantly decreased by the tacrolimus treatment (tacrolimus 49.9 ± 3.4 vs. control 65.3 ± 3.4 pmol·islet^{-1}·60 min^{-1}, $n = 10$, $P < 0.01$), whereas hexokinase activity was not affected (tacrolimus 35.5 ± 2.8 vs. control 34.1 ± 2.1 pmol·islet^{-1}·60 min^{-1}, $n = 10$, not significant; Fig. 3*C*).

DISCUSSION

In the present study, we show that a low concentration of tacrolimus suppresses high glucose-induced insulin secretion from pancreatic islets without affecting insulin content. This inhibitory effect of tacrolimus is the result of reduced ATP production and glycolysis due to decreased glucokinase activity.

We found that 3 nM tacrolimus, a lower concentration than used in previous reports (31, 32, 41), significantly decreased glucose-induced insulin release after 24-h exposure without affecting insulin content, which indicates that reduced insulin release by tacrolimus is not necessarily derived from reduced insulin content. To investigate the mechanism of reduced insulin release by tacrolimus independent of reduced insulin content, we used 3 nM tacrolimus-treated islets. Recommended trough concentrations of tacrolimus in blood are 3–6 ng/ml (3.6–7.2 nM) and 5–15 ng/ml (6.1–18.2 nM) in islet transplantation (36) and in liver transplantation (3), respectively. Ac-

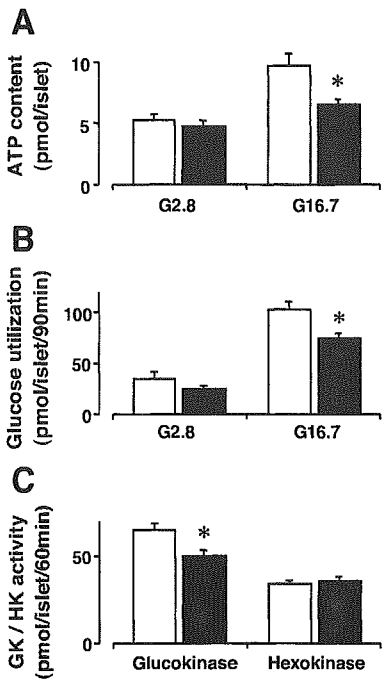

Fig. 3. Metabolic parameters in control (open bars) and tacrolimus-treated islets (solid bars). Islets were cultured with or without 3 nM tacrolimus for 24 h. Cultured islets were preincubated with 2.8 mM glucose for 30 min. *A*: ATP content. After preincubation, islets were incubated in the presence of 2.8 or 16.7 mM glucose. Values represent means ± SE of 10 determinations from several experiments. *B*: glucose utilization. After preincubation, islets were incubated in the presence of 2.8 or 16.7 mM glucose. Values represent means ± SE of 9 determinations from several experiments. *C*: glucokinase (GK) and hexokinase (HK) activities. Values represent means ± SE of 10 determinations from several experiments. $*P < 0.01$ vs. corresponding control.

cordingly, the concentration used in our experiments is comparable to clinically used concentrations.

Chronic exposure to 1 μM tacrolimus for 7 days has been shown not to alter the extent of islet cell apoptosis or necrosis (41), suggesting that apoptosis and necrosis in islets are not increased in our experimental condition. In addition, the inhibitory effect of chronic exposure to 3 nM tacrolimus was almost completely recovered by withdrawal of tacrolimus. For these reasons, the reduced glucose-induced insulin release from 3 nM tacrolimus-treated islets is unlikely to be derived from nonspecific irreversible toxic effects of the agent.

In dynamic experiments, both glucose-induced biphasic insulin release and glucose-induced elevation of NAD(P)H fluorescence were reduced by tacrolimus treatment after 10 min of glucose exposure, suggesting that the reduced insulin release results from reduced glucose metabolism. Accordingly, ATP, the most important metabolic signal in insulin release, was investigated. Because insulin content per islet was not different between control and tacrolimus-treated islets in the present study, the sizes of the stable pool of ATP in an islet, which occurs mainly in the insulin granules, and the diffusible pool, which occurs mainly in the cytosol (4), were not very different. Therefore, the comparison of total ATP content is valid. ATP content in the presence of high glucose was reduced in tacrolimus-treated islets. Mitochondrial ATP production is driven by the H^+ gradient across the mitochondrial membrane generated by transport of high-energy electrons in the respiratory chain. These electrons are derived from NADH and $FADH_2$, which are derived from the TCA cycle in the matrix and/or transferred from the cytosol by the shuttle system. A reduced supply of substrates to mitochondria results in a decreased H^+ gradient across the mitochondrial membrane. Glucose utilization reflects the velocity of glycolysis (24), and NAD(P)H autofluorescence dominantly reflects the redox state of mitochondria (29). Because both glucose utilization and NAD(P)H fluorescence in the presence of high glucose were decreased in tacrolimus-treated islets, the reduced ATP content in these islets may well be attributable to a decreased supply of reduced equivalents to mitochondria.

In pancreatic islets, KIC is oxidized, enhancing ATP production and triggering insulin release (21). However, the mechanism of KIC-induced insulin release is not fully understood, and two distinct mechanisms are proposed. First, KIC, which is converted to acetyl-CoA via a branched-chain α-ketoacid dehydrogenase (BCKDH)-dependent pathway, enters into the TCA cycle and is oxidized (18, 19). Second, KIC, together with endogenous glutamate, is converted to α-ketoglutarate via glutamate-ketoacid transaminase (GKAT), which enters into the TCA cycle and is oxidized (11). Because both BCKDH and GKAT are mitochondrial enzymes, KIC should be metabolized within mitochondria without affecting cytosolic glycolysis. Our results show that mannoheptulose, a glucokinase inhibitor (39), and iodoacetate, a glyceraldehyde-3-phosphate dehydrogenase inhibitor (35), both of which inhibit glycolysis, decreased glucose-induced insulin release but did not affect KIC-induced insulin release. These results are compatible with the notion that KIC is metabolized within mitochondria without affecting cytosolic glycolysis. Accordingly, KIC-induced insulin release was examined to clarify the effect of tacrolimus on mitochondrial metabolism independent of glycolysis. Because tacrolimus reduced glucose-induced insulin release but did not affect KIC-induced insulin release, the decreased glucose metabolism should be derived from reduced glycolysis.

Glucokinase is a rate-limiting enzyme in glycolysis and serves as glucose sensor in pancreatic β-cells (22). Regulation of the enzyme has a central impact on β-cell function, as demonstrated in studies of glucokinase mutations identified in patients with maturity onset diabetes of the young (42). The activity of the enzyme is reduced in tacrolimus-treated islets and presumably plays a primary role in reducing glucose-induced insulin release without a decrease in insulin content by the agent.

Recently, Uchizono et al. (41) reported an effect of chronic exposure to tacrolimus (mainly 100 nM) on the mechanism of insulin release under reduced insulin content. Interestingly, glucose-induced insulin release was found to be more profoundly suppressed than mitochondrial fuel-induced insulin release, a phenomenon clarified by our results. They assert that tacrolimus impairs glucose-induced insulin release and has effects distal to the rise in intracellular Ca^{2+}, which could be explained by a reduced ATP level by the agent, as ATP directly enhances Ca^{2+} efficacy in the exocytotic system of insulin release (9, 33, 40).

Glucokinase activity in pancreatic β-cells is regulated on both translational and posttranslational levels, but the mechanism remains largely unknown (23). Further investigation of suppression of glucokinase activity by tacrolimus may clarify the regulation of glucokinase activity in pancreatic β-cells.

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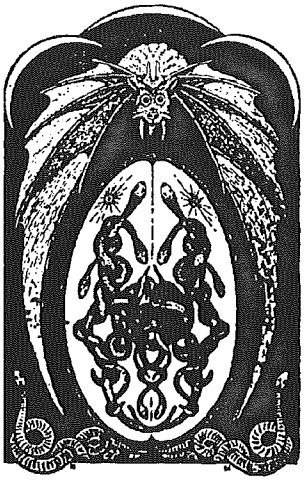

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Chronic exposure to β-hydroxybutyrate inhibits glucose-induced insulin release from pancreatic islets by decreasing NADH contents

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Takehiro, Mihoko, Shimpei Fujimoto, Makiko Shimodahira, Dai Shimono, Eri Mukai, Koichiro Nabe, Razvan Gheorghe Radu, Rieko Kominato, Yo Aramaki, Yutaka Seino, and Yuichiro Yamada. Chronic exposure to β-hydroxybutyrate inhibits glucose-induced insulin release from pancreatic islets by decreasing NADH contents. *Am J Physiol Endocrinol Metab* 288: E372–E380, 2005. First published October 12, 2004; doi:10.1152/ajpendo.00157.2004.—To investigate the effects of chronic exposure to ketone bodies on glucose-induced insulin secretion, we evaluated insulin release, intracellular Ca^{2+} and metabolism, and Ca^{2+} efficacy of the exocytotic system in rat pancreatic islets. Fifteen-hour exposure to 5 mM D-β-hydroxybutyrate (HB) reduced high glucose-induced insulin secretion and augmented basal insulin secretion. Augmentation of basal release was derived from promoting the Ca^{2+}-independent and ATP-independent component of insulin release, which was suppressed by the GDP analog. Chronic exposure to HB affected mostly the second phase of glucose-induced biphasic secretion. Dynamic experiments showed that insulin release and NAD(P)H fluorescence were lower, although the intracellular Ca^{2+} concentration ($[Ca^{2+}]_i$) was not affected 10 min after exposure to high glucose. Additionally, $[Ca^{2+}]_i$ efficacy in exocytotic system at clamped concentrations of ATP was not affected. NADH content, ATP content, and ATP-to-ADP ratio in the HB-cultured islets in the presence of high glucose were lower, whereas glucose utilization and oxidation were not affected. Mitochondrial ATP production shows that the respiratory chain downstream of complex II is not affected by chronic exposure to HB, and that the decrease in ATP production is due to decreased NADH content in the mitochondrial matrix. Chronic exposure to HB suppresses glucose-induced insulin secretion by lowering the ATP level, at least partly by inhibiting ATP production by reducing the supply of NADH to the respiratory chain. Glucose-induced insulin release in the presence of aminooxyacetate was not reduced, which implies that chronic exposure to HB affects the malate/aspartate shuttle and thus reduces NADH supply to mitochondria.

islet; reduced nicotinamide adenine dinucleotide; adenosine 5′-triphosphate

KETONE BODIES ARE PRODUCED IN THE LIVER and are used peripherally as an energy source when glucose is not readily available. Ketosis is seen in various physiological conditions such as fasting, prolonged exercise, and high-fat diet. Hyperketonemia is also caused by absolute or relative insulin deficiency in type 1 diabetic patients or type 2 diabetic patients, respectively (18).

The importance of insulin in control of ketone body production in the liver is well established (18). The function of these substrates in regulating insulin output from pancreatic β-cells is less well understood. In short-term exposure of <60 min, ketone bodies such as D-β-hydroxybutyrate (HB) are oxidized by islet cells and enhance insulin secretion in the presence of stimulatory levels of glucose (4, 10, 15, 28, 29, 31). However, little is known of the effect of prolonged exposure to ketone bodies on pancreatic β-cells and insulin secretion. Zhou et al. (47) have reported that 48-h exposure to HB reduces high glucose-induced insulin secretion from human pancreatic islets without affecting pyruvate dehydrogenase (PDH) activity or PDH kinase activity, although the mechanism of the inhibitory effect is unclear.

The mechanism of glucose-stimulated insulin release from pancreatic β-cells is well documented. Glucose entry into the β-cells accelerates glycolysis and glucose oxidation to increase the ATP content and ATP-to-ADP ratio, which closes the ATP-sensitive K^+ (K_{ATP}) channels. The decrease in K^+ conductance depolarizes the membrane and opens the voltage-dependent Ca^{2+} channels (VDCC). Increased Ca^{2+} influx through VDCCs increases the intracellular Ca^{2+} concentration ($[Ca^{2+}]_i$) to a level that triggers exocytosis of the insulin granules (13). It has been shown that glucose can also promote insulin release K_{ATP} channel independently by increasing Ca^{2+} efficacy in stimulation-secretion coupling, which may be due at least in part to the direct effect of increased ATP derived from glucose metabolism on exocytosis (1, 13). The effect of chronic exposure to ketone bodies in this mechanism is not known, however.

In the present study, insulin release, intracellular Ca^{2+} and metabolism, and the efficacy of Ca^{2+} in the exocytotic system were investigated in HB-exposed islets.

MATERIALS AND METHODS

Materials. NADH, ADP, 2-amino-2-methyl-1-propanol, oxaloacetate, RPMI 1640 medium, alcohol dehydrogenase, aspartate transaminase, malate dehydrogenase, GDPβS, diadenosine pentaphosphate (DAPP), antimycin A, L-β-hydroxybutyrate, lithium acetoacetate, aminooxyacetate, and potassium aspartate were obtained from Sigma Chemicals (St. Louis, MO). Fura PE-3 AM was obtained from Calbiochem (La Jolla, CA). Succinate and HB were purchased from Aldrich (Steimheim, Germany). D-Hydroxybutyrate dehydrogenase was obtained from Toyobo (Osaka, Japan). ^3H_2O, [5-^{3}H]glucose, and [U-^{14}C]glucose were obtained from Amersham (Buckinghamshire, UK). All other reagents are of analytic grade and were obtained from Nacalai Tesque (Kyoto, Japan). Sodium acetoacetate was prepared and incubated for over 16 h at 20°C, from ethyl acetoacetate and NaOH (each 1 M), followed by three successive washes with diethyl

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ether to remove residual ethyl acetoacetate and ethanol formed by hydrolysis as previously described (29). The solution was then flushed with N_2 for 60 min and stored at −80°C.

Acetoacetate determination. Acetoacetate was determined as previously described (12). Briefly, the reaction mixture containing 50 μl of sample, 120 mM triethanolamine (pH 7.0), 375 μM NADH, 1 unit/ml D-hydroxybutyrate dehydrogenase, and 1.25 mg/ml oxamic acid in a total volume of 400 μl was incubated for 15 min at 37°C. The reaction was stopped by heating the mixture at 100°C for 2 min. The concentration was determined by measuring the decrease in absorbance at 340 nm using lithium acetoacetate as standard.

Animals. Male Wistar rats were obtained from Shimizu (Kyoto, Japan). The animals were fed standard lab chow ad libitum and allowed free access to water in an air-conditioned room with a 12:12-h light-dark cycle until used in the experiments. All experiments were carried out with rats aged 8–12 wk. The animals were maintained and used in accordance with the Guidelines for Animal Experiments of Kyoto University.

Islet isolation and culture. Islets of Langerhans were isolated from Wistar rats by collagenase digestion as described previously (6). Islets were cultured for 15 h in RPMI 1640 medium (containing 10% fetal calf serum, 100 IU/ml penicillin, 100 μg/ml streptomycin, and 8.3 mM glucose) with or without β-hydroxybutyrate and acetoacetate at 37°C in humidified air containing 5% CO_2.

Measurement of insulin release from intact islets. Insulin release from intact islets was monitored by use of static incubation and perifusion conditions, as described previously (6), with Krebs-Ringer bicarbonate buffer (KRBB) supplemented with 0.2% BSA and 10 mM HEPES adjusted to pH 7.4 (KRBB medium). For static incubation experiments, cultured islets were preincubated at 37°C for 30 min with 2.8 mM glucose. Groups of five islets were then batch incubated for 30 min in 0.7 ml of KRBB medium with 2.8 and 16.7 mM glucose. At the end of the incubation period, islets were pelleted by centrifugation (15,000 *g*, 180 s), and aliquots of the buffer were sampled.

For perifusion experiments, groups of 20 cultured islets were placed in each of the parallel chambers (400 μl each) of a perifusion apparatus and perifused with KRBB medium at a rate of 0.7 ml/min at 37°C. The medium was continuously gassed with 95% O_2-5% CO_2. Islets were perifused for 30 min to establish a stable insulin secretory rate at the basal level of glucose, and the glucose concentration was then raised to 16.7 mM.

The amount of immunoreactive insulin was determined by RIA, using rat insulin as standard (6). Insulin contents and DNA contents of islets were measured as previously described (8).

Measurement of $[Ca^{2+}]_i$ and reduced pyridine nucleotide fluorescence. For $[Ca^{2+}]_i$ measurement, cultured islets were loaded with fura PE-3 as previously described (7). The islets were immediately placed in a heat-controlled chamber on the stage of an inverted microscope kept at 36 ± 1°C, superfused with KRBB medium containing 2.8 mM glucose for 30 min, and subsequently exposed to the medium containing a high concentration of glucose. These islets were excited successively at 340 and 380 nm, and the fluorescence emitted at 510 nm was captured by a charge-coupled device (CCD) camera (Micro Max 5-MHz System; Roper Industries, Trenton, NJ). The 340-nm (F340) and 380-nm (F380) fluorescence signals were detected every 20 s, and the ratios (F340/F380) were calculated. In vitro calibration was performed as previously described (7). For reduced pyridine nucleotide [NAD(P)H] measurement, the islets without dye were excited successively at 360 nm, and the fluorescence emitted at 470 nm was captured every 20 s (9) by the CCD camera described above. Changes in NAD(P)H fluorescence signal were expressed as percent control values by dividing the signal at a given time by the average signal at 2.8 mM glucose during the last 1 min before stimulation.

Images of $[Ca^{2+}]_i$ and NAD(P)H were analyzed with the Meta Fluor image analyzing system (Universal Imaging, West Chester, PA).

Measurement of insulin release from permeabilized islets. Insulin release from permeabilized islets was determined as previously described (6). After preincubation with 2.8 mM glucose for 30 min, cultured islets were washed twice in cold potassium aspartate buffer (KA buffer) containing 140 mM potassium aspartate, 7 mM $MgSO_4$, 2.5 mM EGTA, 30 mM HEPES, and 0.5% BSA (pH 7.0), with $CaCl_2$ added to a Ca^{2+} concentration of 30 nM. The islets were permeabilized by high-voltage discharge in KA buffer and washed once with the same buffer. Groups of five electrically permeabilized islets were batch incubated for 30 min at 37°C in 0.4 ml of KA buffer with various concentrations of Ca^{2+} and ATP. At the end of the incubation period, permeabilized islets were pelleted by centrifugation (15,000 *g*, 180 s), and aliquots of the buffer were sampled for immunoreactive insulin determination.

Measurement of adenine nucleotide contents. After groups of cultured intact islets were preincubated at 2.8 mM glucose for 30 min, groups of 15 islets were batch incubated in 0.5 ml of KRBB medium containing 2.8 or 16.7 mM glucose at 37°C for 30 min. Incubation was stopped by the addition of 0.1 ml of 2 M $HClO_4$. The tubes were immediately mixed with vortex and sonicated in ice-cold water for 3 min. They were then centrifuged (3,000 *g*, 3 min), and a fraction (0.4 ml) of the supernatant was mixed with 100 μl of 2 M HEPES and 100 μl of 1 M Na_2CO_3. ATP and ADP were assayed by a luminometric method as previously described (8). For measurement of the sum of ATP + ADP, ADP was first converted into ATP by adding 210 μl of solution containing 20 mM HEPES (pH 7.75), 3 mM $MgCl_2$, 1.5 mM phospho*enol*pyruvate, and 2.2 U/ml pyruvate kinase to 70 μl of extracts, with incubation at 37°C for 30 min. The ATP concentration in solutions was measured by adding 200 μl of luciferin-luciferase solution (Turner Designs, Sunnyvale, CA) to a fraction of sample (100 μl) in a bioluminometer (Luminometer model 20e, Turner Designs). For measurement of ATP, the same procedure was performed except that the incubation step was done without pyruvate kinase. The ADP concentration was calculated as the difference between the value of ATP + ADP and ATP from the same sample. To draw a standard curve and to ascertain that the conversion of ADP into ATP was complete, blanks and ADP and ATP standards were run through the entire procedure, including the extraction steps.

Measurement of NADH contents. NADH contents were assayed with an enzymatic cycling system for NADH amplification (17, 45). After preincubation at 2.8 mM glucose, batches of 20 cultured islets were incubated in 50 μl of KRBB medium containing 2.8 or 16.7 mM glucose. After incubation at 37°C for 30 min, 20 μl of solution containing 40 mM NaOH and 0.5 mM cysteine were added to each tube. The tubes were immediately mixed with vortex and sonicated in the ice-cold water for 3 min. They were then centrifuged (3,000 *g*, 3 min), and a fraction (30 μl) of the supernatant was incubated for 20 min at 60°C to destroy any remaining NAD. For the first step in NADH amplification, the reaction mixture containing 30 μl of extract, 100 mM HEPES-NaOH buffer (pH 7.5), 1 mM dithiothreitol, 0.2 mg/ml BSA, 280 mM ethanol, 2 mM oxaloacetate, 16 U/ml alcohol dehydrogenase, and 4 U/ml malate dehydrogenase in a total volume of 100 μl was incubated for 30 min at 30°C. The reaction was stopped by heating the mixture at 100°C for 2 min. In the second step, malate formed in the first step was assayed by incubating 100 μl of sample solution in 50 mM 2-amino-2-methyl-1-propanol-HCl buffer (pH 9.9) containing 0.18 mM NAD, 0.1 mg/ml BSA, 9 mM glutamate, 0.8 unit/ml malate dehydrogenase, and 1 U/ml aspartate transaminase in a total volume of 600 μl for 30 min at 30°C. The NADH formed was measured by fluorometry (Shimazu RF-5000, Kyoto, Japan) at a 340-nm excitation and a 450-nm emission. To draw a standard curve, blanks and NADH standards were run through the entire procedure.

Measurement of mitochondrial ATP production. Mitochondrial suspension from cultured islets was prepared by repeated centrifugation, as previously described (14, 16, 19). First, isolated islets were homogenized in *solution A* consisting of (in mM) 50 HEPES, 100 KCl, 1.8 ATP, 1 EGTA, and 2 $MgCl_2$ and 0.5 mg/ml BSA (electrophoretically homogeneous; pH 7.00 at 37°C with KOH). After precipitation of cell debris and nuclei by centrifugation, the supernatant

was centrifuged more rapidly (10,000 *g*) to obtain a pellet containing the mitochondrial fraction. The precipitation diluted by 200 μl of *solution A* was centrifuged again and rinsed three times in solution consisting of (in mM) 20 HEPES, 3 KH_2PO_4, 1 EGTA, 12 sodium gluconate, 0.3 $MgCl_2$, 148 potassium gluconate, and 4 carnitine and 0.5 mg/ml BSA (electrophoretically homogeneous), adjusted to pH 7.10 with KOH (*solution B*). The mitochondrial fraction in 500 μl of *solution B* was kept on ice until use. To measure ATP production by oxidative phosphorylation, the reaction was started by adding 5 μl of mitochondrial suspension to 495 μl of prewarmed *solution B* (37°C) supplemented with the mitochondrial substrates, 50 μM ADP, and 1 μM DAPP adjusted to pH 7.10. DAPP is a specific inhibitor of adenylate kinase used to measure ATP production by oxidative phosphorylation exclusively. To normalize the mass of the intact mitochondria obtained, ATP production by adenylate kinase, one of the mitochondrial intermembrane kinases, was measured in the presence of ADP but without mitochondrial substrates or DAPP in parallel incubations (19–21). Reaction was stopped by addition of 0.5 μM antimycin A. The samples were cooled to room temperature, and ATP concentration in the solutions was measured by adding luciferin-luciferase solution to each sample with a bioluminometer. ATP production was determined as the ratio of ATP production by oxidative phosphorylation to that by adenylate kinase. To draw a standard curve, blank and ATP standards were added to parallel samples containing the complete incubation mixture except the mitochondrial suspension.

Measurement of glucose utilization and oxidation. Glucose utilization and oxidation were measured, using the previously described method (3) with a slight modification. Cultured islets were preincubated in KRBB medium with 2.8 mM glucose at 37°C for 30 min. For utilization, batches of 30 islets for each condition were incubated at 37°C for 90 min in 150 μl of medium containing 1.5 μCi of [5-^{3}H]glucose. Aliquots of the incubation medium (100 μl) and 20 μl of 1 M HCl were transferred to small tubes and placed into a glass vial containing 0.5 ml of H_2O. The capped vials were incubated overnight at 34°C to vaporize ^3H_2O from the solution. The inner tube was then lifted out, and the disintegrations per minute of water-melting ^3H_2O in the vial were counted. In a parallel incubation, the recovery ratio of ^3H_2O was measured with ^3H_2O. After subtraction of blank disintegrations per minute from sample disintegrations per minute, glucose utilization was calculated with the disintegrations per minute, specific radioactivity of [5-^{3}H]glucose, and recovery ratio of ^3H_2O. For oxidation, all procedures were the same as for utilization except the use of [U-^{14}C]glucose (0.5 μCi/tube) and 0.5 ml of hydroxide of hyamine 10-X (Packard, Meriden, CT) in place of [5-^{3}H]glucose and 0.5 ml of H_2O, respectively. After subtraction of blank disintegrations per minute from sample disintegrations per minute, glucose oxidation was calculated using the disintegrations per minute and specific radioactivity of [U-^{14}C]glucose.

Statistical analysis. Results are expressed as means ± SE. Statistical significance was evaluated by unpaired Student's *t*-test. $P < 0.05$ was considered significant.

RESULTS

Effect of chronic exposure to β-hydroxybutyrate on glucose-induced insulin release from intact islets. Addition of 10 mM HB to RPMI 1640 medium did not alter the H^+ concentration measured by the H^+ electrode (data not shown). Exposure to 1 mM HB for 15 h did not alter 16.7 mM glucose-induced and basal insulin release. However, exposure to 2 mM HB reduced 16.7 mM glucose-induced insulin release but enhanced basal insulin release. No further inhibitory effect on high glucose-induced insulin release and no further enhancement of basal insulin release at 5 mM HB were observed (Table 1, *top*). Insulin release in the presence of 200 μM diazoxide, 16.7 mM glucose, and 30 mM K^+ from HB-cultured islets was also reduced by chronic exposure to 5 mM HB for 15 h (HB 1.22 ± 0.03 vs. control 2.15 ± 0.09 ng·islet^{-1}·30 min^{-1}, $n = 5$, $P < 0.01$). Exposure to 5 mM L-β-hydroxybutyrate for 15 h did not alter 16.7 mM glucose-induced and basal insulin release (Table 1, *bottom*). The DNA contents and insulin contents of islets cultured with 5 mM HB for 15 h were similar to controls (DNA contents: HB 26.6 ± 1.9 vs. control 27.1 ± 1.8 ng/islet, $n = 30$, not significant; insulin contents: HB 34.3 ± 1.9 vs. control 30.6 ± 1.4 ng/islet, $n = 30$, not significant).

In perifusion experiments, insulin release from HB-cultured islets in the presence of 2.8 mM glucose for 5 min before high-glucose exposure was increased compared with control islets (Fig. 1*A*, *inset*; mean value from −5 to 0 min: HB 0.21 ± 0.01 vs. control 0.16 ± 0.01 ng·20 islets^{-1}·min^{-1}, $n = 6$, $P < 0.01$). Insulin release began to elevate ~1 min after exposure to high glucose in both groups of islets (Fig. 1*A*). Insulin release from HB-cultured islets 1–10 min after 16.7 mM glucose exposure was similar to that from control islets. Insulin release from HB-cultured islets for the second and third 10-min period after high-glucose exposure was less than from control islets (Fig. 1*B*; mean value from 20 to 30 min: HB 1.93 ± 0.14 vs. control 2.66 ± 0.07 ng·20 islets^{-1}·min^{-1}, $n = 6$, $P < 0.01$).

Effect of chronic exposure to acetoacetate on glucose-induced insulin release from intact islets. Because acetoacetate (AcA) is spontaneously decarboxylated to acetone, a decline in the concentration of AcA in the medium may occur during the culture period. As expected, decreases in the concentrations of AcA were observed in the medium; however, >65% AcA remained in the medium at the end of the 15-h culture period (Table 2). Exposure to AcA (initial concentration 2–10 mM) for 15 h did not affect 16.7 mM glucose-induced insulin release. However, chronic exposure to high concentrations of AcA (initial concentration 10 mM) augmented basal insulin release (Table 2).

Table 1. *Effect of chronic exposure to D-β-hydroxybutyrate on glucose-induced insulin release from intact islets*

Experimental Condition During Culture	Insulin Release, ng·islet^{-1}·30 min^{-1}	
	Basal (2.8 mM glucose)	Stimulated (16.7 mM glucose)
Control	0.21±0.01	2.89±0.14
1 mM D-HB	0.24±0.01	2.63±0.15
2 mM D-HB	0.29±0.01*	2.46±0.13†
5 mM D-HB	0.31±0.02*	2.18±0.13*
7 mM D-HB	0.29±0.02*	2.24±0.12*
10 mM D-HB	0.26±0.01*	2.15±0.12*
Control	0.19±0.01	2.81±0.11
5 mM L-HB	0.21±0.02	2.99±0.13
5 mM D-HB	0.32±0.03*	2.26±0.15*

Values are means ± SE of 12 (*top*) and 16 (*bottom*) determinations from several experiments. Intact islets were cultured under the conditions indicated for 15 h. After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM glucose for 30 min was determined. *Top:* concentration-dependent effect of D-β-hydroxybutyrate (D-HB) on insulin release. *Bottom:* effect of chronic exposure to D-HB and L-β-hydroxybutyrate (L-HB) on insulin release. *$P < 0.01$ vs. corresponding control (cultured without HB). †$P < 0.05$ vs. corresponding control (cultured without HB).

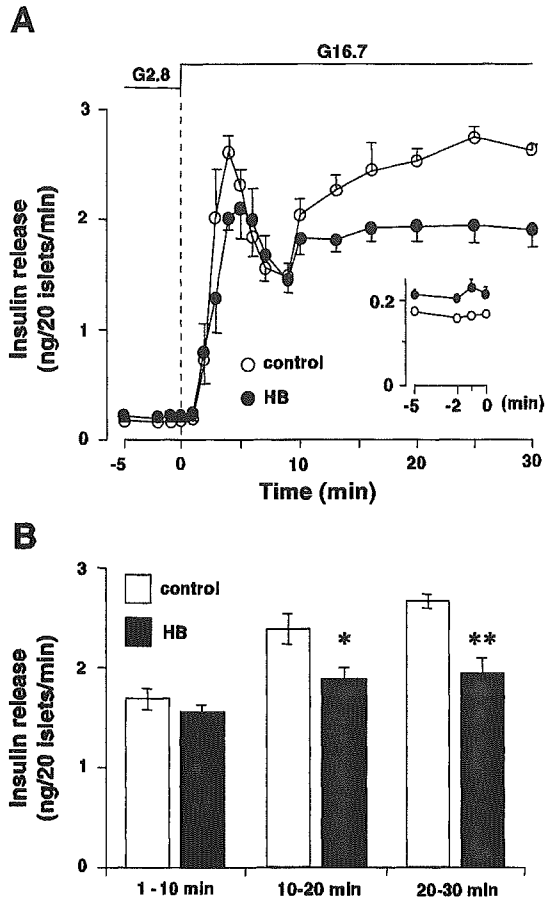

Fig. 1. Effect of D-β-hydroxybutyrate (HB) on biphasic 16.7 mM glucose-induced insulin release from intact islets. Islets were cultured with or without 5 mM HB for 15 h. After 2 groups of islets were perifused for 30 min (−30 to 0 min) with 2.8 mM glucose (G2.8), they were stimulated with 16.7 mM glucose (G16.7; 0–30 min). Values represent means ± SE of 6 determinations in the same experiments. Experiments using the same protocol were repeated 3 times to ascertain reproducibility. *A*: time course of high glucose-induced biphasic insulin release from control and HB-cultured islets. All values in HB-treated islets after 13 min, except those at 16 min, were significantly less than the corresponding control values (at 13 min, $P < 0.05$; at 20, 25, and 30 min, $P < 0.01$). *Inset*: time course of basal insulin release for 5 min (−5 to 0 min) in the same experiment. All values in HB-treated islets from −5 to 0 min were significantly greater than the corresponding control values (at −5 and 0 min, $P < 0.05$; at −2 and 0 min, $P < 0.01$). *B*: average values of insulin release from control and HB-cultured islets during the time indicated. Values are calculated from the data in *A*. *$P < 0.05$ vs. control; **$P < 0.01$ vs. control.

$[Ca^{2+}]_i$ elevation in intact islets induced by a high concentration of glucose. Figure 2*A* shows $[Ca^{2+}]_i$ elevation induced by a high concentration of glucose in control and HB-cultured islets. In the presence of 2.8 mM glucose for 5 min before exposure to high glucose, $[Ca^{2+}]_i$ of HB-cultured islets was higher than in control islets (average from −5 to 0 min: HB 134.8 ± 10.4, $n = 17$, vs. control 86.7 ± 7.6 nM, $n = 13$, $P < 0.01$). However, $[Ca^{2+}]_i$ of HB-cultured islets at 1–10 min and for the second and third 10-min period after exposure to high glucose was similar to that of controls (Fig. 2*B*).

Insulin release from electrically permeabilized islets. In the presence of 5 mM ATP, raising the Ca^{2+} concentration from 30 nM to 3 μM elicited a concentration-dependent increase in insulin release from both electrically permeabilized control and HB-cultured islets. In the presence of 5 mM ATP, insulin release at 30 nM, 100 nM, 1 μM, and 3 μM Ca^{2+} was greater in HB-cultured islets than in control islets (Fig. 3*A*). Augmentation of insulin release at 30 nM Ca^{2+} in HB-cultured islets also was found in the absence of ATP, indicating an effect of the ATP-independent and Ca^{2+}-independent component of insulin release (6) (Fig. 3*B*). When the increment of insulin release in the presence of 30 nM Ca^{2+}, which is due to chronic exposure to HB, was subtracted from the insulin release from HB-cultured islets to evaluate Ca^{2+}-dependent components of insulin release (7), the values at 100 nM, 1 μM, and 3 μM were found to be similar to the values from control islets (Fig. 3*A*). Insulin release without ATP at 30 nM Ca^{2+} from control islets was not affected by 500 μM GDPβS, a stable GDP analog, although that from HB-cultured islets was significantly suppressed by the analog (Fig. 3*B*).

Time course of NAD(P)H fluorescence in intact islets. NAD(P)H fluorescence began to elevate immediately after exposure to high glucose in both groups of islets and reached plateau within 5 min after exposure (Fig. 4). Attenuation of NAD(P)H fluorescence was observed from 2 min in HB-cultured islets (at 2 min: HB 1.16 ± 0.01, $n = 15$, vs. control 1.38 ± 0.03 arbitrary units, $n = 12$, $P < 0.01$). The average of fluorescence during the first, second, and third 10-min period was lower in HB-cultured islets than in control islets (from 0 to 10 min: HB 1.23 ± 0.01, $n = 15$, vs. control 1.50 ± 0.03, $n = 12$, $P < 0.01$; from 10 to 20 min: HB 1.32 ± 0.02, $n = 15$, vs. control 1.68 ± 0.04, $n = 12$, $P < 0.01$; from 20 to 30 min: HB 1.31 ± 0.03, $n = 15$, vs. control 1.68 ± 0.04 arbitrary units, $n = 12$, $P < 0.01$).

Reduced pyridine nucleotide contents and adenine nucleotide contents. NADH contents in the presence of 16.7 mM were greater than in the presence of 2.8 mM glucose in both control and HB-cultured islets. NADH contents of HB-cultured islets in the presence of 2.8 mM glucose and in the presence of 16.7 mM glucose were more and less than control islets, respectively (Table 3, *top*). The ATP contents and the ATP-to-ADP ratio in the presence of 16.7 mM were greater than in the presence of 2.8 mM glucose in both control and HB-cultured islets. The ATP contents and ATP-to-ADP ratio of HB-cul-

Table 2. *Effect of chronic exposure to acetoacetate on glucose-induced insulin release from intact islets*

Experimental Condition During Culture, mM		Insulin Release, ng·islet^{-1}·30 min^{-1}	
Initial	After 15 h	Basal (2.8 mM glucose)	Stimulated (16.7 mM glucose)
Control		0.21±0.02	2.81±0.12
2.0	1.3	0.21±0.02	2.79±0.09
5.0	4.0	0.18±0.01	2.82±0.15
10.0	9.1	0.31±0.02*	3.06±0.20

Values are means ± SE of 15 determinations from 3 experiments. Effect of chronic exposure to various concentrations of acetoacetate on insulin release. Intact islets were cultured under the conditions indicated for 15 h. Initial concentration of acetoacetate in the medium is indicated. At the end of the culture period, acetoacetate concentrations in the medium were determined. After cultured islets were preincubated with 2.8 mM glucose for 30 min, insulin release in the presence of 2.8 and 16.7 mM glucose for 30 min was determined. *$P < 0.01$ vs. corresponding control (cultured without acetoacetate).

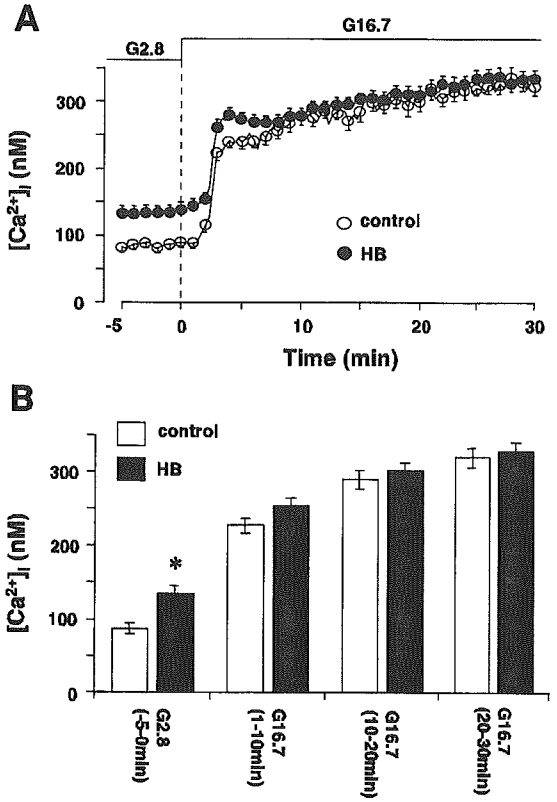

Fig. 2. Intracellular Ca^{2+} concentration ($[Ca^{2+}]_i$) elevation induced by 16.7 mM glucose in control and HB-cultured intact islets. Islets were cultured with or without 5 mM HB for 15 h. After preincubation with 2.8 mM glucose for 30 min, the islets were stimulated with 16.7 mM glucose at *time 0*. *A*: time course of $[Ca^{2+}]_i$ in control and HB-cultured islets. Values represent means ± SE of 13 (control) or 17 (HB) determinations from several experiments. *B*: average values of $[Ca^{2+}]_i$ from control and HB-cultured islets during the time indicated. Values are calculated from the data in *A*. $^*P < 0.01$ vs. control.

tured islets in the presence of 16.7 mM glucose were less than in control islets (Table 3, *bottom*).

ATP production by mitochondria from islets. ATP production by mitochondria from control and HB-cultured islets in the presence of various substrates and inhibitors is shown in Table 4. ATP production by mitochondria from HB-cultured islets in the presence of succinate was decreased compared with that from control islets. Antimycin A, a complex III inhibitor in the respiratory chain, inhibited ATP production dramatically in the presence of succinate in both control and HB-cultured islets. Rotenone, a complex I inhibitor in the respiratory chain, also inhibited ATP production in the presence of succinate in both control and HB-cultured islets. However, in the presence of succinate and rotenone, no significant difference in ATP production was observed in control and HB-cultured islets. Mitochondrial ATP production of HB-cultured islets was similar to that of control islets in the presence of tetramethyl-*p*-phenyldiamine and ascorbate.

Glucose utilization and glucose oxidation. Glucose utilization and glucose oxidation in the presence of 16.7 mM glucose were greater than in the presence of 2.8 mM glucose in both control and HB-cultured islets. Glucose utilization and glucose oxidation of HB-cultured islets in the presence of 2.8 mM glucose were greater than in control islets. However, glucose utilization and glucose oxidation of HB-cultured islets in the presence of 16.7 mM glucose were not different from those of control islets (Table 5).

Insulin release in the presence of aminooxyacetate. Glucose (16.7 mM)-induced insulin release from HB-cultured islets was reduced compared with that from control islets (HB 2.22 ± 0.08 vs. control 2.89 ± 0.16 ng·islet^{-1}·30 min^{-1}, $n = 7$, $P < 0.01$). Aminooxyacetate (AOA), an aspartate aminotransferase inhibitor, reduced 16.7 mM glucose-induced insulin release from both control and HB-cultured islets. However, in the presence of AOA, 16.7 mM glucose-induced insulin release from HB-cultured islets was not different from that of control

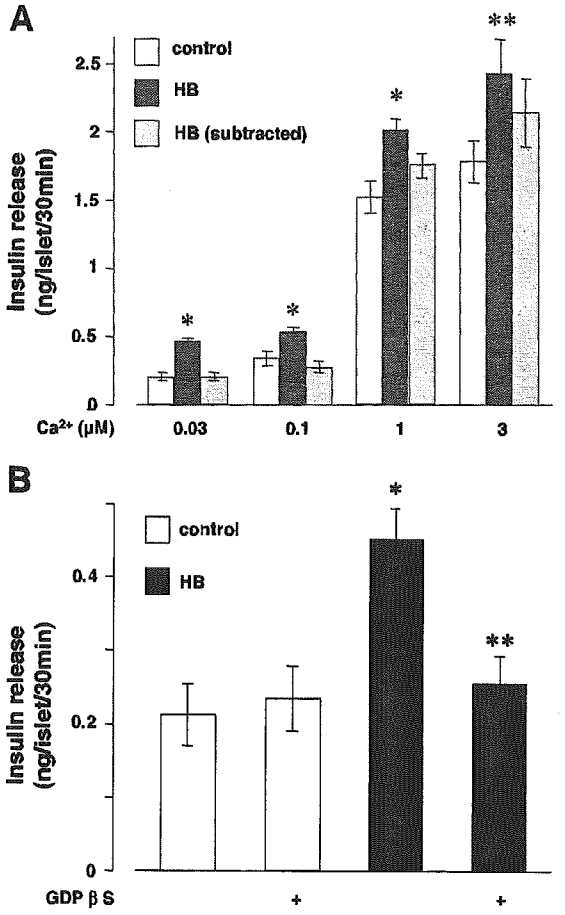

Fig. 3. Effect of HB on insulin release from electrically permeabilized islets in the presence of various concentrations of Ca^{2+} with or without 5 mM ATP. Islets were cultured with or without 5 mM HB for 15 h. After preincubation with 2.8 mM glucose for 30 min, islets were electrically permeabilized and incubated in the indicated medium. *A*: insulin release from control and HB-cultured islets in the presence of 5 mM ATP. Values obtained by subtracting the increment in insulin release in the presence of 30 nM Ca^{2+} with 5 mM ATP due to chronic exposure to HB from the value of insulin release from HB-cultured islets are indicated. Values represent means ± SE of 15 (control) and 16 (HB) determinations from several experiments. $^*P < 0.01$ vs. corresponding control; $^{**}P < 0.05$ vs. corresponding control. *B*: insulin release from control and HB-cultured islets in the presence of 30 nM Ca^{2+} and in the absence of ATP with (+) or without 500 μM GDPβS. GDPβS was present during incubation of permeabilized islets with 30 nM Ca^{2+} and without ATP (for 30 min). Values represent means ± SE of 8 determinations from 2 experiments. $^*P < 0.01$ vs. corresponding control without GDPβS; $^{**}P < 0.01$ vs. HB-cultured islets without GDPβS.

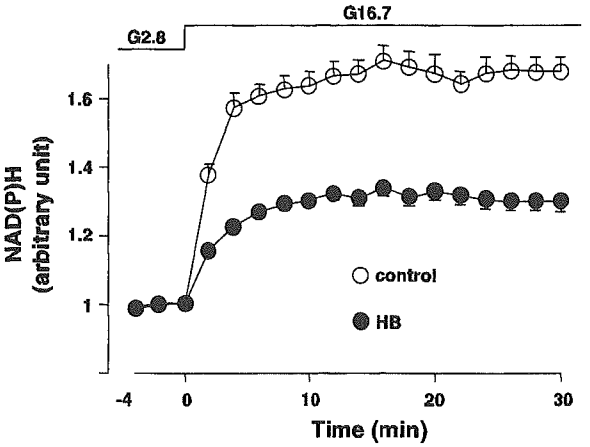

Fig. 4. Time course of NAD(P)H elevation induced by 16.7 mM glucose in control and HB-cultured islets. Islets were cultured with or without 5 mM HB for 15 h. After perifusion with 2.8 mM glucose for 30 min, they were exposed to 16.7 mM glucose at *time 0* for 30 min. NAD(P)H signals were measured at 20-s intervals from −4 to 30 min. Values represent means ± SE of 12 (control) and 15 (HB) determinations from several experiments.

islets (HB 1.98 ± 0.06 vs. control 1.91 ± 0.07 ng·islet^{-1}·30 min^{-1}, n = 7, not significant; Fig. 5).

DISCUSSION

We find that chronic exposure to HB reduces high glucose-induced insulin secretion and augments basal insulin secretion from pancreatic islets. Insulin secretion is reduced by diminished ATP, which is at least partly due to lower ATP production resulting from the reduced supply of NADH to the respiratory chain. Basal insulin secretion is increased due to augmentation of the Ca^{2+}-independent and ATP-independent component of insulin release, in which a GTP-sensitive site may play a role.

In the present study, chronic exposure to >2 mM HB affected insulin secretion, this concentration of HB augmenting glucose-induced insulin acutely in vitro (4, 10, 15, 28, 29, 31) and being frequently observed in serum in various pathophysiological states (18). Acute exposure to the L-isomer was ineffective on insulin release, presumably due to no capacity to oxidize the isomer in islets (4, 28). To exclude nonspecific effects of HB, the chronic effect of the L-isomer was also examined. Because the L-isomer did not affect insulin release, the effect of the D-isomer is presumably linked to its metabolism. Moreover, chronic exposure to AcA, another ketone body, did not affect glucose-induced insulin release. HB is oxidized to AcA by D-β-hydroxybutyrate dehydrogenase, a mitochondrial enzyme, also present in islets (28), producing NADH in mitochondria. Accordingly, alteration of mitochondrial redox potential during culture may be involved in reduced glucose-induced insulin release from HB-cultured islets. Interestingly, acute exposure to exogenous HB or AcA increases or decreases [6-^{14}C]glucose oxidation (28), respectively, which is strongly affected by anaplerosis (41). Anaplerosis plays an important role in determination of the cytosolic level of metabolic factors, including malonyl-CoA (5), NADPH (24), and nonessential amino acids (41) in pancreatic β-cells. The chronic effects of these factors should be taken into account when considering reduced glucose-induced insulin release by HB.

In pancreatic β-cells, intracellular Ca^{2+} and ATP are the most important regulators of insulin secretion, although there may be other independent metabolic signals (2, 25–27, 34). Ca^{2+} and ATP directly affect the exocytotic system and enhance insulin release synergistically in experiments using single β-cells (37, 43) and permeabilized islets (7). In the present study, we have compared $[Ca^{2+}]_i$, intracellular ATP, and

Table 3. *Reduced pyridine nucleotide and adenine nucleotides contents in the presence of basal and stimulated levels of glucose in control and HB-cultured islets*

	Control Islets		HB-Cultured Islets	
	2.8 mM	16.7 mM	2.8 mM	16.7 mM
NADH, fmol/islet	89±7	375±35*	138±11†	256±8*†
ATP, pmol/islet	6.53±0.36	8.26±0.36*	5.91±0.23	6.78±0.35†‡
ADP, pmol/islet	2.21±0.16	1.51±0.08*	1.93±0.09	1.56±0.14‡
ATP-to-ADP ratio	3.06±0.14	5.56±0.17*	3.12±0.10	4.71±0.35*§

Values are means ± SE of 11 (*top*) and 17 (*bottom*) determinations from several experiments. Islets were cultured with or without 5 mM HB for 15 h. After cultured intact islets were preincubated with 2.8 mM glucose for 30 min, they were incubated with 2.8 and 16.7 mM glucose for 30 min. Reaction was stopped, and nucleotides contents were determined. *$P < 0.01$ vs. corresponding islet incubated with 2.8 mM glucose. †$P < 0.01$ vs. corresponding control cultured without HB. ‡$P < 0.05$ vs. corresponding islet incubated with 2.8 mM glucose. §$P < 0.05$ vs. corresponding control cultured without HB.

Table 4. *ATP production by mitochondria from control and HB-cultured islets*

Experimental Conditions	Mitochondrial ATP Production	
	Control islets	HB-cultured islets
1 mM Succinate	2.99±0.20	2.42±0.10*
1 mM Succinate + 1 μM rotenone	2.18±0.16†	1.98±0.12†
20 mg/ml TMPD + 2 mM ascorbate	1.04±0.09	0.98±0.09
1 mM Succinate + 0.5 μM antimycin A	0.09±0.02‡	0.06±0.02‡

Values are means ± SE of 5 determinations from 5 experiments. Islets were cultured with or without 5 mM HB for 15 h. Mitochondrial suspension was obtained from control and HB-cultured islets. Mitochondrial ATP production is indicated as the ratio to ATP production from adenylate kinase, which was determined from the same sample in parallel incubation. TMPD, tetramethyl-*p*-phenyldiamine. *$P < 0.05$ vs. corresponding control cultured without HB. †$P < 0.05$ vs. corresponding succinate alone. ‡$P < 0.01$ vs. corresponding succinate alone.

Table 5. *Glucose oxidation and glucose utilization in control and HB-cultured islets*

	2.8 mM Glucose	16.7 mM Glucose
Glucose utilization		
Control islets	25.1±2.0	84.6±6.7*
HB-cultured islets	39.5±3.9†	90.1±5.1*
Glucose oxidation		
Control islets	3.1±0.4	18.4±3.7*
HB-cultured islets	7.1±0.8†	20.8±1.9*

Values are mean glucose metabolism ± SE (in pmol·islet^{-1}·90 min^{-1}) of 7 (glucose utilization) and 9 (glucose oxidation) determinations from several experiments. Islets were cultured with or without 5 mM HB for 15 h. After cultured intact islets were preincubated with 2.8 mM glucose for 30 min, they were incubated with 2.8 and 16.7 mM glucose for 90 min. *$P < 0.01$ vs. corresponding control incubated with 2.8 mM glucose. †$P < 0.01$ vs. corresponding control cultured without HB.

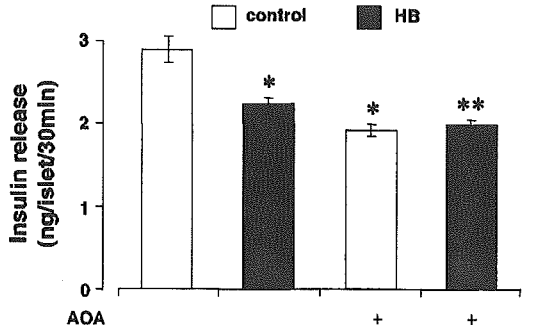

Fig. 5. Glucose (16.7 mM)-induced insulin release from control and HB-cultured islets in the presence of aminooxyacetate (AOA). Islets were cultured with or without 5 mM HB for 15 h. After cultured intact islets were preincubated with 2.8 mM glucose for 30 min, they were incubated with 16.7 mM glucose with (+) or without 5 mM AOA for 30 min. Values represent means ± SE of 7 determinations. *$P < 0.01$ vs. control without AOA; **$P < 0.05$ vs. HB-cultured islets without AOA.

$[Ca^{2+}]_i$ efficacy in the exocytotic system at clamped concentrations of ATP in control and HB-exposed islets.

Chronic exposure to HB decreases insulin release by 16.7 mM glucose during a short period around the peak in the first phase and mostly during the second phase of biphasic secretion, 10 min after exposure. $[Ca^{2+}]_i$ in the presence of 16.7 mM glucose in HB-cultured islets was similar to that in control islets during the second phase. Moreover, insulin release in the presence of diazoxide, high glucose, and a depolarizing concentration of K^+, in which $[Ca^{2+}]_i$ is clamped independently of glucose metabolism (10), is also reduced by chronic exposure to HB. Apparently, $[Ca^{2+}]_i$ does not play a role in the lower response to high glucose in HB-cultured islets. In addition, Ca^{2+} efficacy at clamped concentration of ATP in the exocytotic system was similar in the two groups of islets. On the other hand, NAD(P)H fluorescence was less in the HB-cultured islets in the presence of high glucose. Moreover, both ATP contents and ATP-to-ADP ratio of HB-cultured islets 30 min after exposure to high glucose were reduced compared with control islets. Decreased intracellular ATP reduces Ca^{2+} efficacy in the exocytotic system (7, 37, 43). Therefore, the lower ATP level due to the reduced redox state may play a major role in the attenuation of insulin secretion from HB-cultured islets in response to high glucose.

NAD(P)H autofluorescence dominantly reflects the redox state of mitochondria (32) and is widely used as an index of the intracellular redox state in intact islets (9, 33, 36). However, the method cannot discriminate between NADH and NADPH signals. Therefore, NADH, the supply of which accelerates mitochondrial oxidative phosphorylation, was determined using the enzymatic method. NADH contents of HB-cultured islets were lower in the presence of high glucose, partly accounting for the decreased ATP level in these islets.

To determine whether oxidative phosphorylation in the mitochondrial respiratory chain is suppressed in HB-cultured islets, mitochondrial ATP production was examined in the presence of various substrates and inhibitors. In the presence of succinate, which directly renders electrons to complex II (39), ATP production in HB-cultured islets is less than in controls. The fact that ATP production in both groups of islets in the presence of succinate is partially inhibited by rotenone, a complex I inhibitor (39), indicates that increased NADH derived from succinate also renders electrons to complex I and that electrons from complex I, in addition to electrons from complex II, augment oxidative phosphorylation in the presence of succinate. Tetramethyl-*p*-phenyldiamine with ascorbate renders electrons to cytochrome *C*, which transfers electrons to complex IV (39). The fact that ATP production in the presence of succinate and rotenone and in the presence of tetramethyl-*p*-phenyldiamine and ascorbate is similar in the two groups of islets indicates that the respiratory chain downstream of complex II is not affected by chronic exposure to HB. Accordingly, the decrease in ATP production in the presence of succinate may be due to decreased NADH content in the matrix of mitochondria derived from succinate.

NADH in the matrix of mitochondria is produced in a reaction catalyzed by isocitrate dehydrogenase, α-ketoglutarate dehydrogenase, and malate dehydrogenase in the TCA cycle, and by PDH (23, 38). Malate dehydrogenase also participates in the malate/aspartate shuttle, which carries NADH produced in cytosol to the matrix of mitochondria (22, 40). PDH activity is not affected by chronic exposure to HB in human islets (47). The fact that glucose utilization and glucose oxidation were not affected indicates that isocitrate dehydrogenase, α-ketoglutarate dehydrogenase, and PDH, which catalyze reactions accompanying production of NADH and CO_2, were not affected by chronic exposure to HB. Malate dehydrogenase catalyzes reactions that produce NADH without production of CO_2 that are involved in both the TCA cycle and malate/aspartate shuttle. AOA (5 mM) reduced glucose-induced insulin release from rat islets without affecting glucose oxidation, because AOA does not affect the TCA cycle and inhibits the malate/aspartate shuttle by blocking transamination reactions (30). Glucose-induced insulin release in the presence of AOA was not reduced by chronic exposure to HB, suggesting that chronic exposure to HB affects the malate/aspartate shuttle.

With the use of electrically permeabilized islets incubated at low Ca^{2+} without ATP, it was revealed that augmentation of basal insulin release from HB-cultured islets does not necessarily require elevation of $[Ca^{2+}]_i$, although $[Ca^{2+}]_i$ was higher in HB-cultured islets in the presence of basal levels of glucose than in control islets. This augmentation is not due to nonspecific leakage of insulin and to slow washout of insulin, since it was suppressed by the GDP analog, which is also compatible with the fact that GTP augments insulin release in the exocytotic process directly and Ca^{2+} independently (35).

Postprandial insulin release is inhibited in vivo in the starved state (11, 42, 46) and in type 2 diabetic patients with ketosis (44). Although the present results suggest suppressed insulin release in such pathophysiological conditions, these results may well not be found in vivo, as elevation of basal insulin secretion is not observed in the fasting state.

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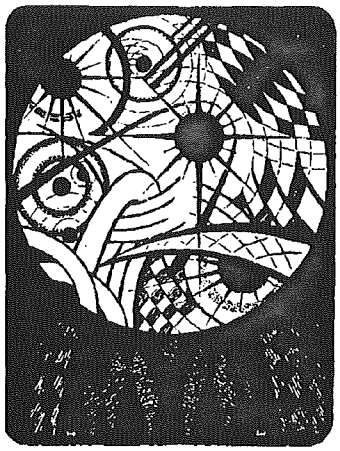

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IGT with fasting hyperglycemia is more strongly associated with microalbuminuria than IGT without fasting hyperglycemia

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Abstract

Previous studies have established that impaired glucose tolerance (IGT) patients with fasting hyperglycemia (IGT/FH: fasting plasma glucose (FPG) level 6.1–7.0 mmol/l and 2 h PG level of 7.8–11.1 mmol/l) exhibit higher insulin resistance than those with isolated IGT (FPG level <6.1 mmol/l and 2 h PG level of 7.8–11.1 mmol/l), but the association with microalbuminuria has not been determined. Here, we evaluate the prevalence of microalbuminuria in non-diabetic Japanese males 20–70 years of age. The subjects were classified into four groups based on the results of OGTT: normal glucose tolerance (NGT: $n = 71$), impaired fasting glucose (IFG: $n = 24$), isolated IGT ($n = 36$), and IGT/FH ($n = 23$). A urinary albumin-to-creatinine ratio (ACR) from 30 to 300 μg/mg creatinine was counted as microalbuminuria. The prevalence of microalbuminuria was higher in subjects with IGT/FH than in subjects with isolated IGT (26% versus 14%). Logistic regression analysis showed microalbuminuria to be more significantly associated with IGT/FH (OR = 3.82, 95% CI 1.09–13.36) than with isolated IGT (OR = 1.75, 95% CI 0.50–6.17). While insulin resistance (HOMA-IR) in isolated IGT was not significantly different from that in NGT, insulin resistance in IGT/FH was significantly higher ($P < 0.01$). Regression analysis of ACR in IGT showed a significant correlation with insulin resistance ($P = 0.012$). Accordingly, microalbuminuria is more strongly associated with IGT/FH than with isolated IGT, most likely due to the higher insulin resistance.

Keywords: Microalbuminuria; Impaired glucose tolerance; Fasting hyperglycemia; Type 2 diabetes; Insulin resistance

1. Introduction

Microalbuminuria reflects widespread vascular damage due to generalized endothelial dysfunction [1–3], and is a predictor of all-cause mortality and

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cardiovascular morbidity in diabetic patients as well as in non-diabetic subjects [4–7]. Microalbuminuria has a significant association with impaired glucose tolerance (IGT), and both increasing insulin resistance and hypertension are thought to be major factors in its development [8–11].

As impaired fasting glucose (IFG) has been identified as a pre-diabetic condition by 1998 WHO diagnostic criteria [12], IGT can be divided into IGT without fasting hyperglycemia (isolated IGT; FPG level: <6.1 mmol/l, 2 h PG level: 7.8–11.1 mmol/l) and IGT with fasting hyperglycemia (IGT/FH; fasting plasma glucose (FPG) level: 6.1–7.0 mmol/l, 2 h plasma glucose (PG) level: 7.8–11.1 mmol/l). Several studies have shown that subjects with IGT/FH have higher insulin resistance than those with isolated IGT [13–15]. However, the association between IGT/FH and microalbuminuria has not been determined. In the present cross-sectional study, we have evaluated the prevalence of microalbuminuria in Japanese males with varying degrees of glucose intolerance.

2. Material and methods

2.1. Subjects

We consecutively recruited Japanese male subjects undergoing 75 g OGTT for closer evaluation if they had a family history of diabetes, positive urine glucose test, or >5.5% of HbA_{1c} level at initial examination for regular medical check-up at Kyoto University Hospital and its affiliated hospitals between 1991 and 2001. OGTT was performed within 3 months of initial examination. Subjects with clinical proteinuria or occult blood in urine were excluded because of possible progressive diabetic nephropahty or other renal diseases. Subjects with FPG level higher than 11.0 mmol/l and those who had received any treatment for diabetes or hypertension also were excluded. The study was designed in compliance with the ethics regulations of the Helsinki Declaration.

Standard oral glucose tolerance test was administered according to National Diabetes Data Group recommendations [16], which require the subject to fast overnight for 10–16 h. Blood samples for the determination of blood glucose were drawn 0 and 120 min after oral administration of 75 g glucose. Blood samples for measurement of HbA_{1c}, insulin, total cholesterol, HDL-cholesterol, and triglyceride were drawn after an overnight fast.

Based on the results of 75 g OGTT, the subjects were classified into the following five groups: normal glucose tolerance (NGT), impaired fasting glucose (IFG), isolated IGT, IGT/FH, and DM. The NGT group included subjects with FPG level less than 6.1 mmol/l and 2 h PG level less than 7.8 mmol/l (n = 71). The IFG group included subjects with FPG level between 6.1 and 7.0 mmol and 2 h PG level less than 7.8 mmol/l (n = 24). The isolated IGT group included subjects with FPG level less than 6.1 mmol/l and 2 h PG level between 7.8 and 11.1 mmol/l (n = 36). Subjects with FPG level between 6.1 and 7.0 mmol/l and 2 h PG level between 7.8 and 11.1 mmol/l comprised the IGT/FH group (n = 23). Subjects with FPG level not less than 7.0 mmol/l and/or 2 h PG level not less than 11.1 mmol/l comprised the DM group (n = 149).

2.2. Measurements

Weight and height were measured in light clothing without shoes, and body mass index (BMI, kg/m^2) was calculated. Blood pressure was checked at the brachial artery in supine position after 10 min rest. Plasma glucose was measured by glucose oxidase method using a Hitachi Automatic Clinical Analyzer 7170 (Hitachi, Tokyo, Japan). Serum insulin was measured by radioimmmunoassay (Dainabot, Tokyo, Japan). Serum total cholesterol, HDL-cholesterol, and triglyceride level were measured on a Hitachi 7170 (Hitachi, Tokyo, Japan), as reported previously [17].

As the measure of insulin resistance, the index of insulin resistance by homeostasis model assessment (HOMA-IR) was used (FPG (mmol/l) × fasting insulin (mU/l)/22.5) [18]. HOMA-IR correlates well with measurements obtained by glucose clamp and minimal model studies [19,20].

Early morning first voided urine samples were collected. Urine albumin was measured by a commercial immunoprecipitation assay (LX2000, Eiken, Tokyo, Japan) with sensitivity of 3.0 μg/ml and intra- and inter-assay coefficients of variation of less than 10%. Urinary creatinine was determined